MULTIPEST ECONOMIC THRESHOLDS ON SNAP BEANS

BY

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A DISSERTATION PRESENTED TO THE GRADUATE SCHOOL
OF THE UNIVERSITY OF FLORIDA IN
PARTIAL FULFILLMENT OF THE REQUIREMENTS
FOR THE DEGREE OF DOCTOR OF PHILOSOPHY

UNIVERSITY OF FLORIDA

1986

ACKNOWLEDGEMENTS

I wish to express my sincere gratitude to many people for their support and guidance:

Dr. V. H. Waddill, my advisor and committee chairperson, for his guidance, encouragement, suggestions, and assistance throughout my Doctor of Philosophy program at the University of Florida.

Dr. J. R. Strayer, co-chairperson, for his constructive suggestions, encouragement, and providing materials for computer work whenever I needed them.

Dr. R. McSorley, for serving on my committee, encouragement, suggestions, and invaluable assistance with statistical data analysis.

Dr. K. Pohronezny, for serving on my committee, encouragement, suggestions, and assistance with data analysis.

Dr. S. H. Kerr, for his guidance and constructive suggestions during my Ph.D. program.

I am indebted to Dr. R. D. Berger for his advice, helpful suggestions on disease progress analysis, and allowing me to use his computer programs.

The invaluable technical assistance and friendship of Nancy Shivers, Diane Putnal, W. (Hank) Dankers, Jorge Parrado, James Reynolds, John Sarvich, and Ingeborg Stough during the long and difficult field and laboratory hours are appreciated. I am also indebted to William Meyers, Joyce Francis, Jeanette Viola, and all staff at Homestead TREC for their

help and moral support. I owe a lot to my fellow students for their suggestions and sense of humor.

Sincere thanks are extended to the Malawi government and USAID for the scholarship.

Finally, the most special and loving acknowledgement is owed to my wife Abigail, for her love, encouragement, and patience throughout the program. I would like to end with my sincere thanks to Olivia and Kondwarri, my children, for putting-up with my absence.

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of the University of Florida in Partial Fullfillment
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May, 1986

Chairman: Dr. Van H. Waddill
Major Department: Entomology and Nematology

During 1984 and 1985, a number of greenhouse and field experiments were carried out at the Tropical Research and Education Center at Homestead in Dade County, Florida, to determine the effect of manual defoliation, Meloidogyne incognita, bean rust, and various other nematodes on 'Sprite' snap beans (Phaseolus vulgaris L.). Treatments consisted of total defoliation (100%), 0%, 25%, 50%, and 75% defoliation at various plant growth stages; 0, 10, 100, 1,000, 10,000 and 100,000 M. incognita eggs and juveniles per pot, fungicide sprays which included bitertanol at 7-day intervals, mancozeb tank-mixed with sulfur at 4-5-day, 7-day, and 14-day intervals respectively, and soil fumigation with metam-sodium at 0, 47, 94, 187, 281, and 374 L/ha and a separate one at 935 L/ha. Experiments were conducted with each series of treatments as well as with combinations of two or more types of treatments simultaneously.

Manual defoliation caused the highest snap bean yield losses at full-bloom and pod-set both in the field and the greenhouse experiment. Snap bean yield loss was observed at the 25% defoliation level in both experiments. Total defoliation resulted in the highest yield losses.

Yield was negatively correlated to Meloidogyne incognita (Kofoid and White) Chitwood population levels when plants were grown in pots and the nematodes were used alone. Yield was also inversely related to nematode population levels when manual defoliation occurred on nematode inoculated plants. Yield loss was observed on plants grown in soil inoculated with 10 eggs and juveniles per pot.

The bean rust disease, Uromyces phaseoli (Pers.) Wint., was manipulated by fungicide sprays. It was observed that plants with the highest disease severity gave the lowest yield whereas plants which were virtually disease free had the highest yield. Generally, fungicide sprays increased yields. In some cases yield increases were not high enough to pay for the extra cost of fungicide sprays.

Soil funigation with metam-sodium increased yields slightly. The optimum metam-sodium application rate was 187 L/ha.

Yield was affected most by the bean rust disease when defoliation, metam-sodium and the disease were used simultaneously.

CHAPTER I
INTRODUCTION

Beans, Phaseolus vugaris L., are the major protein source for many people in the world, especially in developing countries (Yamaguchi, 1978). Consequently beans are considered an important crop in the tropics, subtropics, and warm temperate areas of the world (Zaumeyer and Meiners, 1975). Zaumeyer and Meiners (1975) stated that the leading world bean producers were Brazil, Mexico, and the United States of America (U.S.A.) in descending order. Beans are grown for fresh market, processing, and dry seed. In some countries the leaves are also used as a vegetable.

Snap beans are known by various names in different areas. These names include French beans, green beans, pole beans, string beans, and wax beans (Yamaguchi, 1978). Snap beans are grown in many states in the U.S. where 132,720 metric tons (mt.) were harvested for fresh-market consumption from 35,847 hectares (ha.) in 1981 (Anon., 1981). In the same crop year, 671,640 mt. of snap beans were harvested for processing from 93,324 ha. (Anon., 1981). The gross value for snap beans in 1981 was $199,282,000 in the U.S. (Anon., 1981).

Florida is the largest producer of fresh-market snap beans in the U.S., producing nearly 40% of the crop (Anon., 1972, 1982; Rose, 1975; Ware and McCollum, 1980). In the 1981-82 production year, Florida produced 60,600 mt. of bush and pole beans from 37,206 ha. (Anon., 1982). Southeast Florida is the major producing area for snap beans in

the state with the greatest production in Dade County (Anon., 1982). Gadsden, Marion and Palm Beach counties and parts of the west central area also produce some snap beans (Anon., 1982). Rose (1975), Ware and McCollum (1980), and Anon. (1982) reported that the winter demand for fresh market snap beans in population centers to the north of Florida is usually met by supplies from the southern districts of Florida.

Whereas Florida is the largest producer of fresh market snap beans, Wisconsin is the leading producer of snap beans for processing (Kobriger and Hagedorn, 1983). Michigan, New York, and Oregon also produce more snap beans for processing than Florida (Kobriger and Hagedorn, 1983; Ware and McCollum, 1980).

Snap bean production has some inherent pest problems (Anon., 1982; Rose, 1975). Fresh market snap bean yield, however, increased from 31 cwt/acre in the 1947-1952 period to 37 cwt/acre in the 1967-1972 period, in the U.S., despite these problems (Anon., 1972). This increase in yield has been ascribed to the advent of synthetic organic pesticides during and after World War II. In Florida, snap bean yields were, however, on the decline during the same period (Rose, 1975). The decline was associated mainly with adverse weather conditions (Anon., 1982). Galvez et al. (1977) and Vargas (1980) stated that insect pests, such as leafminers, leaffrollers, corn earworm, Mexican bean beetle, and others cause tremendous losses in bean yield. Many of these insects feed on the leaves reducing the photosynthetic tissue of the plant. These pests have been more-or-less controlled by insecticide sprays (Acland, 1971; Iraneta and Rodrigez, 1983).

Bean rust, Uromyces phaseoli, (Pers.) Wint., is one of the most important diseases of beans in many bean producing areas of the world

(Acland, 1971; Cook, 1978; Crispin and Dongo, 1962; Iraneta and Rodrigez, 1983; Martinez, 1983; Schwartz et al., 1979; Stoezer and Omunyin, 1983; Vargas, 1980, Zaumeyer and Thomas, 1957). Other major bean disease include anthracnose (Colletotrichum lindemuthianum Sacc. and Magn.), angular leaf spot (Isariopsis griseola Sacc.), halo blight (Pseudomonas phaseolicola (Burk.) Dows.), common blight (Xanthomonas phaseoli (E. F. Smith)), and bean common mosaic virus (Acland, 1971; Allen, 1983; Martinez, 1983; Stoetzer and Omunyin, 1983). Martinez (1983) stated that root rots caused by Macrophomina phaseolina (Tassi) Goid.,, Sclerotium rolfsii Sacc., Rhizoctonia solani Kuhn, Pythium spp. and Fusarium spp. were among the most important diseases of beans.

Fifty-seven races of the bean rust fungus, Uromyces phaseoli, have been reported in the U.S. (Laundon and Waterston, 1965). The number of races of U. phaseoli is, however, not fixed, due to controversy on what constitutes a physiologic race of a pathogen (Crispin and Dongo, 1972; Davidson and Vaughan, 1963; Groth and Shrum, 1971; McMillan, 1972).

Uromyces phaseoli has been reported to reduce the translocation of photosynthetic products from the foliage to the roots and developing seeds (Daly, 1976; Livne, 1962; Montalbini, 1973; Zaki and Durbin, 1965). The reduction of photosynthetic products translocation is exacerbated by increased water loss through the damaged leaf cuticle despite a decrease in transpiration (Vargas, 1980). Water loss increases as infection becomes more severe. Infection by U. phaseoli predisposes bean plants to other pathogens such as Pythium spp., Rhizoctonia spp. P. phaseolicola, C. lindemuthianun and many others (Vargas, 1980).

Root-knot nematodes, Meloidogyne spp., also infect bean plants. Meloidogyne spp. are most prevalent in light sandy soils with good drainage and moderately warm temperatures (25-30°C) (Crispin et al., 1976). Roberts and Boothroyd (1984), however, stated that M. incognita (Kofoid and White) Chitwood is more common in southern states of the U.S. and M. hapla Chitwood is commonly found in northern states. Meloidogyne spp. reportedly limit the production of beans by interfering with nitrogen fixation by Rhizobium spp. and causing root galls (Ngundo, 1977; Sharma and Guazelli, 1982; Singh et al. 1981 a). Meloidogyne incognita has been observed to predispose beans to Fusarium wilt (Singh et al., 1981 b). Severe root-knot nematode infections may lead to 50-90% yield loss (Freire and Ferraz, 1977; Ngundo, 1977; Varon and Galvez, 1974).

Control of these pests and diseases has been based on chemical pesticides and cultural methods (Acland, 1971; Allen, 1983; Carvalho et al., 1981; Martinez, 1983; Rhoades, 1976; Robbins et al., 1972; Shorey and Hall, 1963; Stoetzer and Omunyin, 1983; Villamonte, 1965; Yoshii, 1977; Zaumeyer and Meiners, 1975). The use of resistant varieties and flooding has been part of the management strategies for bean rust and root-knot nematodes (Crispin et al., 1976; Martinez, 1983; Ngundo, 1977; Singh et al., 1981a; Vieira, 1967).

One of the problems research scientists are confronted with, in crop pest management, is the development of multi-pest threshold levels to be used in determining the type of management strategies and the extent to which these pest complexes have to be controlled so that maximum yield is obtained with minimum disruption of the environment. Pesticides are, however, the main means of controlling pest complexes on

many crops (Allen, 1983; Stoetzer and Omunyin, 1983). Metcalf (1975) reported that repeated use of pesticides has sometimes led to a decline in acreage or yield of the crop. As a consequence of this subtle decline in crop yield due to repeated pesticide use, an integrated pest management (IPM) approach has been advocated (Huffaker and Smith, 1980; Waddill et al., 1981). IPM aims at better understanding of the significance of biological, ecological, and economic processes in the production of the crop, and the population dynamics of the pest complex, their predators and parasites, and other factors affecting the system in the field (Huffaker and Smith, 1980).

Research was undertaken during the period 1984-1985 to investigate the effects of manual defoliation, root-knot nematode population levels, and bean rust on snap bean yield. These factors were used separately, in combinations of two or more of them together, and were studied as stated in order to determine their threshold levels singly and when they occurred simultaneously.

Specific experiments were conducted to determine

1. the effect of defoliation on yield of snap beans under field and open greenhouse conditions,
2. the effect of Meloidogyne incognita population levels on snap bean yield,
3. the effect of bean rust, Uromyces phaseoli on snap bean yield,
4. the effect of defoliation and M. incognita population levels on snap bean yield,
5. the effect of defoliation and several nematode genera on snap bean yield,

6. the effect of defoliation, nematodes and bean rust on snap bean yield, and
7. the effect of inoculation system on the establishment of M. incognita in beans (Phaseolus vulganis L.).

CHAPTER II
LITERATURE REVIEW ON DEFOLIATION AND THE IDENTIFICATION AND CONTROL OF ROOT-KNOT NEMATODES (MELOIDOGYNE SPP.) AND BEAN RUST (UROMYCES PHASEOLI [PERS.] WINT.)

Introduction

Bean insect pests such as leafminers (Liriomyza spp.), bollworms (Heliothis armigera Hbn.), and leaf rollers (Urbanus proteus L.) as well as diseases including rust (Uromyces phaseoli [Pers.] Wint.), web blight (Thanatephones cucumeris [Frank] Donk) and angular leaf spot (Isariopsis griseola Sacc.) not only destroy plant foliage but also cause physiological damage in some cases (Acland, 1971; Galvez et al., 1977). Root-knot nematodes, Meloidogyne spp., cause prolific galls on the root system of plants which may lead to the following above-ground symptoms: incipient wilting, stunted growth, and chlorotic leaves, often with burnt out edges (Agudelo, 1980). A combination of these organisms on beans usually leads to great losses in yield. Control of these pest problems has been mainly by the use of chemical pesticides (Acland, 1971; Agudelo, 1980).

This review is a summary of the problems encountered in the identification of root-knot nematodes and bean rust and the use of simulated leaf damage on beans.

Simulated Leaf Damage on Crop Plants

Bean plants are susceptible to defoliation by insects, diseases, hail, moisture stress, and mechanical injury resulting from farm

machinery. Sometimes defoliation is initiated by chemical defoliants to facilitate harvesting (McGregor et al., 1953). In most cases defoliation occurs due to pest problems and adverse environmental conditions. To reduce defoliation by pests, preventative spray programs are usually followed (Greene and Minnick, 1967). These sprays are usually applied regardless of the anticipated crop loss. Hence, it would be desirable to determine the relationship between defoliation levels and yield losses to maximize the efficiency and rationale for spray treatments.

A wide range of yield losses due to artificial defoliation has been observed on various bean cultivars (Edje and Mughogho, 1976a, 1976b; Edje et al., 1973, 1976; Garvez et al., 1977; Greene and Minnick, 1967; Hohmann and De Carvalho, 1983; Vieira, 1981; Waddill et al., 1984). Edje et al. (1973, 1976), Edje and Mughogho (1976a, 1976b), Vieira (1981) and Waddill et al. (1984) manually defoliated indeterminate bean cultivars. Waddill et al. (1984) reported that complete defoliation when only primary leaves were present reduced yield by about 65% and repeated weekly defoliation of 50% resulted in 34% yield loss. Vieira (1981) reported that 66% leaf area removal during the flowering and pod formation stages was detrimental to yield. Galvez et al. (1977) observed that 100% defoliation at formation of the first trifoliate leaves decreased yields of the bean cultivars ICA-Guali and Porrillo-Sentetico by 34% and 49% respectively. Greene and Minnick (1967) indicated that yield reduction in snap beans begins somewhere between 33% and 50% defoliation when defoliation occurs in the prebloom and bloom stages, respectively. Hohmann and DeCarvalho (1983) reported that removal of 25, 50, 75, and 100% of the leaf area at the pod formation stage reduced

yield by 11, 20, 20, and 70% respectively. At the same levels of leaf area reduction, defoliation at initiation of flowering decreased yield by 18, 12, 19, and 55% respectively. At the formation of the third trifoliate leaves only total defoliation affected the yield.

Kalton et al. (1945) and Weber (1955) reported that 50% and 75% leaf removal in soybeans had little effect on yield when defoliation occurred at the prebloom stage. Significant yield losses were, however, observed when plants were heavily defoliated at the bloom or pod set stages (Begum and Eden, 1963, 1964; Camery and Weber, 1953; Kalton et al., 1945; McAlister and Krober, 1958). Todd and Morgan (1972) observed significant yield reduction on soybeans with 33, 67, and 100% leaf removal at 2 wk, 4 wk, and 8 wk after first bloom. Wilkerson et al. (1984) reported that all defoliations on 'Florunner' peanuts resulted in lower stem weight to length ratios and lower pod numbers and weights. It was observed that defoliation altered the normal partitioning of photosynthates between plant parts in peanuts. Wit (1983) reported that during the most sensitive period (July) in the Netherlands, 60% defoliation induced a maximum yield reduction of 35% in Brussel sprouts. He also noted that when partial defoliation was carried out 15 wk after transplanting or later, no effect on yield was observed. Douglas et al. (1981) observed a grain yield reduction of 77% in corn when complete defoliation was carried out at silking. Grain yield losses decreased with delay in defoliation toward maturity. Less severe defoliations, however, resulted in smaller reductions in yield. Generally, grain yield was tolerant of post-silking defoliation and yield losses exceeding 20% were recorded only after 67% of the leaves were removed. Defoliation action thresholds for tomato for the prebloom and postbloom

stages have been established at 30% and 50% respectively (Keularts, 1980). Extensive research on artificial defoliation effects on tomato has been conducted by various writers (Keularts, 1980; Wolk et al., 1983). The effect of leaf removal has also been studied on cotton (Ludwig, 1926), grain sorghum (Stickler and Pauli, 1961) and wheat and oats (White, 1962; Wotmack and Thurman, 1962).

Nematodes Associated with Beans

Many nematodes have been found in and around the roots of beans (Agudelo, 1980; Allen, 1983). Among the nematodes associated with beans, root-knot nematodes, Meloidogyne spp., are the most important in tropical and subtropical regions (Agudelo, 1980; Allen, 1983). Table 1 shows the nematode species associated with beans in various bean producing areas (Agudelo, 1980; Ayala and Ramirez, 1964; Bridge, 1973; Bridge et al., 1977; Castillo and Litsinger, 1978; Caveness et al., 1975, Feakin, 1973; Hague, 1980; Sinclair and Shurtleff, 1975; Singh and Farrell, 1972).

Of the four main species of Meloidogyne, M. hapla Chitwood has a more northerly distribution than M. arenaria (Neal) Chitwood, M. incognita (Kofoid and White) Chitwood, and M. javanica (Treub) Chitwood which are cosmopolitan in warmer regions (Allen, 1983; Roberts and Boothroyd, 1984). The distribution of the other nematode genera is shown on Table 1.

Occurrence and Importance of Root-knot Nematodes

The most common species of root-knot nematodes are Meloidogyne arenaria, M. incognita, M. hapla, and M. javanica. Meloidogyne arenaria, M. incognita, and M. javanica occur worldwide warmer regions whereas M. hapla has a more northerly distribution (Agudelo, 1980; Allen, 1983;

TABLE 1. Nematodes commonly found in association with roots of beans.

Species	Distribution	Reference
Meloidogyne arenaria (Neal) Chitwood	Cosmopolitan, tropical to warm temperate regions	Agudelo, 1980; Castillo and Litsinger, 1980
M. hapla Chitwood	N. Europe, Japan, U.S.A., Canada, & warmer regions of Africa and Middle East	Agudelo, 1980; Sinclair and Shurtleff, 1975
M. incognita (Kofoid & White) Chitwood	Cosmopolitan, tropical to warm temperate regions	Agudelo, 1980
M. javanica (Treub) Chitwood)	Cosmopolitan, tropical to warm temperate regions	Agudelo, 1980; Sinclair & Shurtleff, 1975
Pratylenchus brachyurus (Godfrey) Filipjev	Cosmopolitan	Agudelo, 1980; Bridge, 1973
Aphelenchoides spp.	Nigeria	Agudelo, 1980; Bridge et al., 1977
Rotylenchulus reniformis Linford & Oliveira	W. Africa, U.S.A., Indonesia, Philippines	Agudelo, 1980; Ayala & Ramirez, 1964; Singh & Farrell, 1972
Helicotylenchus spp.	Cosmopolitan	Agudelo, 1980; Bridge, 1973; Hague, 1980
Criconemella spp.	Widespread	Agudelo, 1980; Feakin, 1973

TABLE 1. Continued.

Species	Distribution	Reference
Belonolaimus spp.	Southeastern U.S.A.	Agudelo, 1980; Feakin, 1973; Sinclair & Shurtleff, 1975
Trichodorus spp.	Widespread	Agudelo, 1980; Feakin, 1973
Xiphinema spp.	Widespread	Agudelo, 1980; Caveness et al., 1975; Feakin, 1973

Roberts and Boothroyd, 1984). Meloidogyne arenaria is rarely encountered in association with beans. Meloidogyne incognita and M. javanica frequently occur simultaneously on beans (Ngundo, 1977; Saka, 1982; Santacruz, 1983; Singh et al., 1981a). The most serious threat to bean production is M. incognita (Ngundo, 1977; Singh et al., 1981a; Sharma and Guazelli, 1982). These nematodes may cause yield losses of 50 to 90% during severe infections (Freire and Ferraz, 1977; Varon and Galvez, 1974).

The limitation on bean by root-knot nematodes has been reported to be due to extensive root-galling and interference with nitrogen fixation by Rhizobium spp. (Agudelo, 1980), as well as with water and nutrient uptake. Root-knot nematode infestations often lead to abbreviated root systems (Agudelo, 1980; Franklin, 1978). Above-ground symptoms of root-knot infections include incipient wilting, chlorotic above-ground plant parts, and stunted growth (Agudelo, 1980).

Epidemiology and Life Cycle of Meloidogyne spp.

Meloidogyne spp. are most abundant in light sandy soils with adequate drainage and temperatures of 25°-30°C (Crispin et al., 1976). Root-knot nematodes are spread by irrigation and flood waters, by vegetative propagation of plant parts in soil contaminated with eggs and juveniles, which adhere to farm implements, animals, and man (Agudelo, 1980; Caveness, 1967; Crispin et al., 1976; Steadman et al., 1975; Vieira, 1967; Villamonte, 1965; Walker, 1965). The length of survival of root-knot nematodes in the soil varies with the stage of development, soil type, moisture, temperature, soil aeration, and length of the fallow period (Navarro and Barriga, 1970; Villamonte, 1965; Walker, 1965).

The life cycle of Meloidogyne spp. has several developmental stages (Taylor and Sasser, 1978). The adult female lays eggs in a gelatinous matrix. The first-stage juvenile develops and molts within the egg. What emerges from the egg is actually the second-stage juvenile, hence the general belief that root-knot juveniles grow between a series of three molts into adult males and females (Agudelo, 1980). Root-knot nematode eggs are oval or ellipsoidal and may be concave on the side. They measure 30-52 x 67-128 µm (Thorne, 1961). These eggs are usually protected from dehydration by a gelatinous matrix secreted by the female (Franklin, 1978; Taylor and Sasser, 1978).

The juvenile stages are vermiform, have a stylet about 10 µm long and have an overall length of 375-500 µm and a width of 15 µm (Robbins et al., 1972; Taylor and Sasser, 1978). Males are cylindroid and measure 0.03-0.36 x 1.2-1.5 mm. The males lack a bursa. Adult females are pyriform and usually pearly white (visible in roots without magnification). The females measure 0.27-0.75 x 0.40 x 1.30 mm and have a soft cuticle (Franklin, 1978; Taylor, 1965; Walker, 1965). The life cycle of root-knot nematodes may take 17-57 days, depending on the soil temperature and the host plant (Tyler, 1933; Taylor and Sasser, 1978).

Infection by and pathogenesis of Meloidogyne spp. are affected by plant age, plant susceptibility, population size and environmental factors (Brodie and Dukes, 1972; Gilvonio and Ravines, 1971; Nemec and Morrison, 1972; Sosa Moss and Torres, 1973). Second stage juveniles of Meloidogyne spp. enter the plant root system within 2 days after inoculation and migrate inter and/or intracellularly through the cortex into the stele (Dropkin, 1980; Ngundo and Taylor, 1975 b). The juvenile inserts its head into the vascular system of the root to obtain

nutrients from the plant. Plant cells in the vicinity of the nematode juvenile increase both in number and size (hyperplasia and hypertrophy), causing the characteristic giant cells (synctia) (Dropkin, 1980; Taylor and Sasser, 1978). The giant cells usually form near the juvenile's head by the fusion and enlargement of plant cells in response to nematode feeding. These giant cells eventually become apparent in the form of galls on the root system. Injury to plant root systems usually becomes apparent 10 days after infection. Five to six weeks after infection, epidermal cells of the roots collapse after females have deposited eggs near the outer root surface (Ngundo and Taylor, 1975a).

Control of Root-knot Nematodes, Meloidogyne spp.

The economic importance of plant-parasitic nematodes is commonly assessed by the use of soil fumigants (Mountain, 1965). Usually, an inverse relationship between yield and nematode numbers is expected (Sasser et al., 1968). The relationship between yield and nematode counts is not always inverse (Robbins et al., 1972). In many bean-producing regions, nematicides are extensively used on a preventative basis (Agudelo, 1980). The world farming community has many nematicides available depending on supply and legal registration. These nematicides include dicholoropropene-dichloropropane (DD), ethylene dibromide (EDB), phenamiphos, methyl bromide, aldicarb, metam-sodium, and DBCP (Jimenez, 1976; Parisi et al., 1972; Rhoades, 1976; Sosa Moss and Wrihs, 1973). In these operations, no attempt is made to eradicate nematodes (Thomason and McKenry, 1975). These nematicide applications are aimed at reducing the nematode populations by 80-90% in the upper 40-60 cm of the soil and are considered adequate to provide economic control (Thomason and McKenry, 1975).

Crop rotation has been used to reduce nematode numbers in bean fields (Agudelo, 1980). Beans are planted once every 2 or 4 years in rotation with a crop such as corn, which is not particularly susceptible to many nematodes parasitic on beans. Cover crops such as marigold (Tagetes minuta), rattle box (Crotalaria spectablilis), or hairy indigo (Indigofera hirsuta) have been used for this purpose (Eguiguren et al., 1975; Navarro and Barriga, 1970; Rhoades, 1976; Zaumeyer and Thomas, 1957). Other cultural practices used to reduce nematode numbers include long fallow periods, deep plowing, and flooding (Crispin et al., 1976; Vieira, 1967).

There are many bean cultivars resistant to M. incognita (Blazey et al., 1964; Christie, 1959; Fassuliotis et al., 1970; Hartman, 1968; Ngundo and Taylor, 1974; Rhoades, 1976; Varon and Galvaz, 1974; Wester et al., 1958). In some cases resistance to M. incognita is broken by simultaneous infection of M. incognita and M. javanica (Ngundo, 1977). Ngundo (1977) reported that seven bean lines were resistant to M. incognita and M. javanica when they occurred simultaneously.

Identification of Root-knot Nematodes

Maggenti (1981) and Taylor and Sasser (1978) state that root-knot nematodes were first described by Berneley in England in 1855. M. incognita was studied, independently, in the U.S. by Neal and Atkinson in 1889 (Maggenti, 1981). Maggenti (1981) reported that Neal indicated that root-knot nematodes occurred in Florida before 1805. During these early studies, Meloidogyne spp. were described under the species names Heterodera marioni or H. radicicola (Maggenti, 1981). Chitwood and Chitwood (1950), as a result of their work on the taxonomy of root-knot nematodes, placed them under the genus Meloidogyne. Chitwood and Chitwood recognized five species of Meloidogyne and one subspecies.

Esser et al. (1976), however, recognized 35 species in this genus. Dickson (unpublished) reported that more than 50 species of Meloidogyne were identified. The number of species in this genus fluctuates due to various identification procedures used and discovery of new species each year.

Root-knot nematode speciation is based on perineal patterns, the distance between stylet knobs and the dorsal esophageal gland opening, the second-stage juvenile morphology, chromosome number, electrophoresis, and host range (Maggenti, 1981; Taylor and Sasser, 1978). Host differentials are also used to separate races of the same species (Taylor and Sasser, 1978).

Meloidogyne incognita is the most widely distributed species of root-knot nematode, comprising 52% of a world collection (40°N to 33°S) in areas where annual temperatures are normally within the 18-30°C range (Taylor and Sasser, 1978). This species has four host races as follows: race 1 does not infect 'Deltapine' 16 cotton, 'NC95' tobacco, and 'Florunner' peanuts; race 2 does not infect 'Deltapine' cotton, and 'Florunner' peanuts; race 3 does not infect 'NC95' tobacco and 'Florunner' peanuts; and race 4 does not infect 'Florunner' peanuts only. All four races infect 'California Wonder' pepper, 'Charleston Grey' watermelon and 'Rutgers' tomato (Taylor and Sasser, 1978). Meloidogyne incognita has a very extensive host range and frequently coexists with M. javanica (Dickson, unpublished; Santacruz, 1983). Meloidogyne javanica is the second most widely distributed species, forming 31% of a world collection (Taylor and Sasser, 1978). Meloidogyne javanica has no known host races but exhibits variation in chromosome numbers. M. hapla and M. arenaria comprised 8 and 7% of a world

collection respectively (Taylor and Sasser, 1978). These two species are known to have 2 host races each (Dickson, unpublished).

The Importance of Bean Rust

Bean rust is known to occur whenever beans are grown (Vargas, 1980). Bean rust is the most important bean disease in Central and South America (Augustin et al., 1972; Crispin and Dongo, 1962; Makram et al., 1973; Zaumeyer and Meiners, 1975). Bean rust has been reported to reduce the yield of snap beans in New Zealand, Egypt, and Australia (Ballantyne, 1974; Makram et al., 1973; Yen and Brien, 1960). Yields of dry beans have been lowered by infections of bean rust in Kenya and Turkey (Mukumya, 1974; Rudolph and Baykal, 1978).

Although the occurrence of bean rust was characterized as sporadic in the U.S. (Harter et al., 1935), Vargas (1980) reported yield losses as high as 40-80% in the U.S. are caused by this disease. Brazil is reported to incur losses of 35-50% due to bean rust infections (Vargas, 1980).

Bean rust was reported to be responsible for the bulk of the yield losses in Navy beans in Michigan (Andersen, 1975). The disease was reported to be troublesome in snap bean fields of North Dakota and Minnesota (Meiners, 1977). Zaumeyer and Meiners (1975) reported that prior to 1945, bean rust was a major disease in irrigated fields in Colorado, western Nebraska, Wyoming and Montana. In their review, Zaumeyer and Meiners (1975) reported bean rust was no longer a problem in those areas, although it was still occasionally important in fall snap bean crops along the Atlantic seaboard and in winter crops grown in Florida.

Identification and Etiology of the Pathogen

Bean rust is caused by the fungus Uromyces phaseoli [Pers.] Wint. (= U. phaseoli typica (Reben) Wint. = U. appendiculatus [Pers.] Unger). The fungus was first described in Germany in 1795 (Cook, 1978). The pathogen is an autoecious macrocyclic rust fungus (Kolmer et al., 1984; Laundon and Waterston, 1965). This pathogen is parasitic on the leguminous genera Dolichos, Phaseolus, and Vigna (Laundon and Waterston, 1965). The fungus is transmitted generally through wind-borne uredospores. Uredospores are rusty orange in color and ellipsoidal to obvoidal in shape, 20-30 x 20-26 μm in measurement (Laundon and Waterston, 1965).

The aecial and pycnial stages are rare in U. phaseoli (Harter et al., 1935). Harter et al. (1935) did not observe any aecia or pycnia of this fungus in the field. In Queensland, Ogle and Johnson (1974) did not report seeing mature aecia or pycnia of U. phaseoli. The absence of aecia on U. phaseoli under field conditions has also been reported in Maryland (Marcus, 1952). The aecial stage of this fungus has, however, been observed and reported in New York and Virginia (Fromme and Wingard, 1921; Jones, 1960). Both aecia and pycnia were reported to occur on field grown beans in North Dakota by Venette et al. (1978).

Fromme and Wingard (1918) and Harter et al. (1935) reported that telia form under unfavorable conditions for the development of the pathogen such as low temperatures, decreased host vigor, and increased host resistance. The propensity of the pathogen to form telia was suspected to be an innate character of the fungal isolate (Harter et al., 1935). It has been reported that teliospores do not occur in Florida (Townsend, 1939). Consequently, Townsend (1939) suggested

uredospores blown in from the north serve as primary inocula in Florida. Later, Kidney (1980) observed telia in Alachua and Dade counties, contrary to Townsend's findings.

Uredospores overwinter in infected crop debris and trellis poles (Davison and Vaughan, 1963). These overwintered uredospores are known to initiate the disease cycle in the next growing season in Oregon and Maryland (Davison and Vaughan, 1963; Marcus, 1952). In Florida, colder temperatures than those normal for that state are apparently required for the uredospores to be viable for relatively long periods (Davison and Vaughan, 1963).

Disease development is frequently initiated by uredospores under natural conditions. The uredospore produces a germ tube upon germination. An appressorium which molds itself into the stomatal ledge is formed when the germ tube gets in touch with the stoma (Mendgen, 1973). An infection peg develops, from the appressorium and pushes the guard cells apart until the fungal cytoplasm is transferred into the substomatal vesicle (Vargas, 1980). Enzymes, lipid bodies, and glycogen particles are contained in the vesicle (Mendgen, 1973). The fungus develops infection hyphae and haustoria as it proceeds inter-cellularly in the host tissue (Mendgen and Heitefuss, 1975; Vargas, 1980).

The bean rust fungus may complete its life cycle within 10-15 days after inoculation (Yarwood, 1961). Uredospores are released passively from pustules and disseminated by farm implements, insects, animals, and wind currents (Yarwood, 1961; Zaumeyer and Thomas, 1957).

Symptoms

Apparently, bean rust is primarily a foliar disease which occasionally occurs on pods, stems and branches (Fromme and Wingard, 1918; Laundon and Waterston, 1965; Vargas, 1980).

The uredia (uredinia) are the major diagnostic sign of the pathogen (Fromme and Wingard, 1918). In a susceptible reaction, symptoms of bean rust first appear on the lower leaf surface as minute, whitish, slightly raised spots about 5-6 days after infection. These spots enlarge to form mature reddish-brown pustules which rupture the epidermis and obtain a diameter of 1-2 mm, 10-12 days after infection (Vargas, 1980). The uredia reach a diameter of 5 mm by the 14th day after infection (Rey and Lozano, 1961). The size of the uredia varies depending on environmental conditions as well as the host. The uredia may appear powdery due to uredospores protruding from them (Fromme and Wingard, 1918). Uredia often appear on both leaf surfaces. The uredia are frequently surrounded by chlorotic halos and eventually by rings of secondary and tertiary sori (Zaumeyer and Thomas, 1957). As infection progresses, the leaf becomes debilitated and the chlorotic areas surrounding pustules coalesce, while tissue ramified by the fungus remains green, apparently, as a result of starch accumulation (Wang, 1961). Severe rust infections may cause premature abscission. Bean rust rarely causes small, circular necrotic lesions on pods (Kucharek and Simone, 1980).

Rust infection has been reported to cause increased respiratory rates in susceptible hosts (Daly et al., 1961). Twenty-four hours after infection starch accumulation decreases sharply in the vicinity of the fleck. Accumulation of starch at the perimeter of the lesion, however, resumes 96-120 hr after infection. The quantity of starch in this area decreases at the time the fungus sporulates (Schipper and Mirocha, 1969). Rust infections cause leakage of ions, amino acids, and sugars in susceptible plant leaves (Hoppe and Heitefuss, 1974a). Hoppe and Heitefuss (1974b) presented evidence that rust infection caused damage

to chloroplast membranes. Raggi (1978) reported decreased photosynthetic rates in rust-infected plants.

Epidemiology of the Disease

Fromme and Wingard (1921) reported that since rust rarely attacks pods directly, resulting losses are insidious and difficult to assess. Yield losses are, however, more likely to be severe when plants are infected during the prebloom and flowering stages of development (Almeida et al., 1977; Costa, 1972; Crispin et al., 1976; Nasser, 1976; Wimalajeewa and Thavam, 1973; Yoshii and Galvez, 1975). Early infection of some bean varieties can lead to almost complete crop loss in some seasons (Fromme and Wingard, 1921; Howland and MaCartney, 1966; Townsend, 1939). Townsend (1939) indicated that total loss of the entire crop due to rust has occurred in Florida.

The variability in the prevalence of bean rust seasonally and geographically is partly due to environmental conditions (Augustin et al., 1972; Gonzalez, 1976; Harter and Zaumeyer, 1941; Harter et al., 1935; Schein, 1961). Infection by U. phaseoli is favored by prolonged periods (8-18 hours) of at least 95% RH and moderate temperatures (15-27°C) (Augustin et al., 1972; Gonzalez, 1976, Schein, 1961). The optimum temperature for uredospore germination was reported to be 14.5°C whereas the optimum temperature for infection was 17°C (Harter et al., 1935). Crispin et al. (1976), Schein (1961), and Zaumeyer and Thomas (1957), however, reported that any temperatures below 15° may retard fungal development. Day length and light intensity are also important factors for the development of the bean rust fungus (Harter and Zaumeyer, 1941).

Fifty-seven races of U. phaseoli have been identified in the U.S. (Stavely, 1984). Laundon and Waterston (1965) reported 35 races of U. phaseoli. Races 1 and 2 were identified from specimens obtained from Washington, D.C. and California (Harter et al., 1935). Twenty races of U. phaseoli were differentiated according to their reaction on seven bean cultivars (Harter and Zaumeyer, 1941). Fisher (1952) isolated 10 races from the Rocky Mountain states and Maryland. Race 31 was identified from New Mexico and race 32 from Maryland (Sappenfield, 1954; Zaumeyer, 1960). Hikida (1961, 1962) isolated and identified races 33 and 34 in Oregon. Race 35 was isolated by McMillan (1972) from the bean cultivar Dade, which was bred for resistance to previously known races of U. phaseoli in Florida. McMillan (1972) reported that races 1, 2, 5, 9, 10, 11, and 35 occur in Florida.

There is tremendous variability in the reaction pattern of U. phaseoli races (Groth and Shrum, 1977). In many areas where several races occur, there is usually one race which is greatly predominating (Fisher, 1952).

Uromyces phaseoli races have also been identified outside the U.S. Thirty-one races have been identified in Mexico (Crispin and Dongo, 1962), 10 races in Colombia (Zuniga and Victoria, 1975), 46 races in Brazil (Pereira and Chaves, 1977), 12 races in Puerto Rico (Lopez, 1976), 4 races in Nicaragua, 5 races in Honduras (Vargas, 1969, 1970), 7 races in Guatemala (Vargas, 1970), 5 races in El Salvador (Vargas, 1971), 4 races in Peru (Guerra and Dongo, 1973), 11 races in Costa Rica, 11 races in Australia, and 8 races in East Africa (Ballantyne, 1974, 1975; Fisher, 1952; Ogle and Johnson, 1974).

Control of the Disease

Cultural control measures of this disease include crop rotation and removal of old plant debris (Vieira, 1967). Reduced plant density and planting date adjustment for specific production areas may reduce rust incidence (Vargas, 1980). Resistant varieties of beans have been used for the control of rust (Augustine et al., 1972; Ballantyne, 1974; Coyne and Schuster, 1975; Crispin et al., 1976; Madriz and Vargas, 1975; Meiners, 1974; Rivera, 1977; Rodriguez, 1976).

Fungicidal sprays are usually recommended to help manage bean rust. Since bean rust reduces yields more severely when infection occurs before flowering than when infection is initiated after flowering, fungicidal sprays are, therefore, more effective if applied during early plant development (Yoshii and Galvez, 1975). Of the older fungicides, sulfur dusts have given relatively good control (Ballantyne, 1975; Harter et al., 1935; Zaumeyer and Thomas, 1957). Sulfur is usually applied at the rate of 25-30 kg/ha every 7-10 days. Generally, protectant fungicides fail in areas where rainfall is frequent because deposits are washed off too soon. Other preventative chemicals applied at schedule similar to that of sulfur are chlorothalonil (225 g/ha), maneb (4-5 kg/ha), and mancozeb (3-4 kg/ha) (Costa, 1972; Crispin et al., 1976; Hilty and Mullins, 1975; Vieira, 1967; Wimalajeewa and Thavam, 1973).

Plantvax (Oxycarboxin) is somewhat therapeutic when sprayed 20 to 40 days after planting at the rate of 1.8-2.5 kg/ha (Costa, 1972; Frenhani et al., 1971; Hilty and Mullins, 1975). McMillan et al. (1982) reported effective control of bean rust when bean plants were sprayed weekly with bitertanol or triadimefon. These fungicides are not registered for use on beans at this time. While certain fungicides are

effective against bean rust, their use is regulated by their estimated cost-effectiveness. Thus, Issa and Arruda (1964) cited by Vargas (1980) concluded that chemical control of bean rust was not economically practical in Brazil. This conclusion may apply to most tropical bean-producing areas. The use of fungicides in highly mechanized agricultural systems, such as the U.S., may be economically feasible provided registration conditions are met.

Interaction of Root-knot Nematodes and Other Pathogens

Increased incidence of plant diseases has been reported to be associated with the presence of root-knot nematodes (Brodie and Cooper, 1964; Carter, 1975a,b; Cauquil and Shepherd, 1970; Minton et al., 1975; Morrell and Bloom, 1981; Norton, 1960; Reynolds and Hanson, 1957; Schuster, 1959; Thomason et al., 1959; Van Gundy et al., 1977). Carter (1975), Cauquil and Shepherd (1970), Norton (1960), Reynolds and Hanson (1957), and White (1962) reported increase incidence of soreshin of cotton (Rhizoctonia solani Kuhn), root rot (Pythium debaryanum Hesse) and Fusarium wilt (Fusarium oxysporum Schlecht) when Meloidogyne incognita (Kofoid and White) Chitwood was present. Increased incidence of southern blight, Sclerotium rolfsii Sacc., was observed in soybeans infested with root-knot nematodes (Minton et al., 1975). The interaction of M. incognita and bacterial wilt (Corynebacterium fluccumfaciens (Hedges) Dows.) was reported on beans by Schuster (1959). Van Gundy et al. (1977) reported the enhancement of the development of R. solani in the presence of exudates from galls caused by M. incognita. Morrell and Bloom (1981) reported a significant increase in the percentage of Fusarium wilt occurrence and vessel infection at 21°C in the presence of M. incognita in tomato. Meloidogyne-Fusarium synergism was also observed

in cowpea (Thomason et al., 1959). Interaction of root-knot nematodes is not limited to nematode-fungus or nematode bacterium complexes. Meloidogyne incognita has also been reported to interact with other nematodes (Thomas and Clark, 1983). Thus, Thomas and Clark (1983) reported that early season counts of M. incognita and Rotylenchulus reniformis Linford and Oliveira were positively correlated with later counts of the same nematode, but negative correlations were found between early M. incognita and subsequent R. reniformis, and between early R. reniformis and subsequent M. incognita counts. The authors suggested that a competitive interaction existed with each species capable of inhibiting the other and becoming the dominant population.

Bookbinder and Bloom (1980) reported the interaction of Meloidogyne spp. with bean rust, Uromyces phaseoli (Pers.) Wint. They observed that U. phaseoli and M. incognita were synergistic in suppressing shoot and root weights of beans. Meloidogyne incognita infections reduced uredial diameter of U. phaseoli significantly. It was observed that simultaneous inoculations of U. phaseoli and M. incognita resulted in reduced mean numbers of galls per gram of root tissue. Similar effects were observed when U. phaseoli was inoculated first. Meloidogyne incognita numbers were consequently reduced by 62% in rusted plants. This reduction in nematode numbers was probably due to suppressed translocation of photosythates to the roots (Bookbinder and Bloom, 1980). Egg hatch was, nevertheless, not affected by the fungus.

CHAPTER III
THE EFFECT OF MANUAL DEFOLIATION ON SNAP BEAN
(PHASEOLUS VULGARIS L.) YIELD

Introduction

Snap beans, Phaseolus vulgaris L., are defoliated by leaf eating insects, diseases, mechanical injury, and adverse weather conditions (Agudelo, 1980; Costa and Rossetto, 1972; Ruppel and Idrobo, 1962; Schoonhoven and Cardona, 1980; Vargas, 1980). Thus, an understanding of the yield-loss relationship between pest infestations and a crop is essential for the development of an integrated pest management program. Much information on the relationship between a crop and pest infestations has been obtained by simulating pest attack through manual defoliation of plants (Edje et al., 1972, 1973, 1976; Edje and Mughogho, 1976a, b; Galvez et al., 1977; Greene and Minnick, 1967; Vieira, 1981; Waddill et al.; 1984). Manual defoliation is not a precise simulator of pest defoliation (Ruesink and Kogan, 1975); however, it provides a good estimate of the host-pest relationship. To minimize or avoid defoliation by pests, producers often resort to preventive pesticide applications on their crop (Greene and Minnick, 1967). These pesticide applications are a form of insurance on the crop when little knowledge on the pest damage-yield loss relationship is available.

Beans are defoliated by a wide range of leaf-eating insects including leafminers (Liriomyza spp.), cabbage looper (Trichoplusia ni

(Hub.)), leafroller (Urbanus proteus L.), and beetles (Systates spp.). Pohronezny et al (1978) reported that Liriomyza spp. were considered by many local farmers in Dade County, Florida, as the most serious pest on their crops. Farmers expect yield loss as long as same leaf damage is observed, but Harris (1974) showed that leaf consumption by pests does not necessarily result in yield reduction.

Greene and Minnick (1967) reported that snap bean yield was not significantly reduced until more than 33% of the leaf surface was removed during blooming. It was later observed that snap bean plants tolerated up to 66% defoliation if damage occurred before flowering (Greene, 1971). Vieira (1981) found that 66% leaf loss on an indeterminate bean cultivar reduced yield when defoliation occurred during flowering and pod formation. At the first trifoliate leaf stage, Galvez et al. (1977) found that total (100%) defoliation decreased yield of two bean cultivars by 34 and 49% respectively. Total defoliation of plants when only primary leaves were present resulted in yield reduction of 65% on pole beans in Dade County (Waddill et al. 1984).

Kalton et al. (1945) and Weber (1955) reported that up to 75% leaf removal in soybeans had little effect on yield if plants were defoliated prebloom. Defoliation in the bloom and pod formation stages, however, resulted in significant yield losses (Begum and Eden, 1963, 1964; Camery and Weber, 1953; Kalton et al., 1945; McAlister and Krober, 1958). Todd and Morgan (1972) reported significant yield reductions on soybeans with 33%, 67% and 100% defoliation at 2 wk, 4 wk, and 8 wk after first bloom. Research on the effects of defoliation on crop yield has also been conducted on tomato, cotton, corn, wheat, oats, and other grain crops (Dungan, 1930; Keularts, 1980; Ludwig, 1926; Stickler and Pauli, 1961; White, 1946; Wolk et al. 1983; Womack and Thurman, 1962).

This study on snap beans (Phaseolus vulgaris L., 'Sprite') was conducted to determine the plant growth stage most sensitive to defoliation, and the effects of defoliation on yield.

Materials and Methods

Two defoliation experiments were conducted in the summer and fall, 1984 in the greenhouse and field, respectively. Bush snap beans (Phaseolus vulgaris L. 'Sprite') were grown at the Tropical Research and Education Center in Homestead, Dade County, Florida. The crops were grown on Rockdale soil (pH ca. 7.8). The greenhouse and field experiments were planted on 25 June 1984, and 24 October 1984, respectively. Fertilizer (8:16:16) was applied at the rate of 448 kg/ha according to the University of Florida Extension recommendations (Stall and Sherman, 1983). Benlate(R) (550g ai/ha) was applied fortnightly for control of certain diseases and sprays of Trigard(R) (150 g/ha) were applied at the same frequency for leafminer (Liriomyza spp.) control. Ambush(R) (40 g ai/ha) or Pydrin(R) (250 g ai/ha) was applied at 14-day intervals for cowpea curculio (Chalcodermus aeneus Boh.) control.

Defoliation levels investigated were total (100%), 25%, 50%, and 75%. An undefoliated control was included. Plants were defoliated at the primary leaf stage, first trifoliate leaf, third trifoliate leaf, flower bud formation, full bloom, and pod set. Beans were harvested on 8-20 August 1984 and 18-26 December 1984. The harvest was not graded since cowpea curculio feeding damage to pods was extensive.

Greenhouse experiment

Rockdale soil (3030 L) was fumigated with Dowfume(R) MC2 (681 g) on a cement slab under a tightly sealed polyethylene sheet. Number two

(7.5 L) pots were filled with 6.4 L soil and placed on a corrugated bench 0.91 m high. Six seeds were planted in each pot and thinned to three after emergence. A plot consisted of three pots with three plants/pot. The crop was irrigated twice a day using an automatic time-controlled, water-mist-producing overhead system. The foliage was removed from the distal end of the petiole and the correct number of leaves removed at a particular growth stage was determined by leaf counts.

The treatments were replicated four times and randomized in a complete block. Fresh weights of pods were determined. Yield loss (percentage) was computed from the untreated control yield at each growth stage. The dollar economic value was computed by extrapolating plot data to a per hectare basis. Plot yield data were subjected to analysis of variance and regression using the general linear models procedure of SAS (Ray, 1982).

Field experiment

The herbicides Treflan(R) (841 g ai/ha) and Dual(R) (1.7 kg ai/ha) were applied to the site prior to planting. Plots were kept as weed-free as possible by mechanical cultivation. Plots were three rows wide (0.91 m row spacing) and 6 m long. Seeds were mechanically planted at 8-10 cm spacing within the row. The crop was irrigated using an overhead

sprinkler system. The foliage was removed from the distal end of the petiole and the correct proportion of the foliage removed was determined from leaf counts.

Ambush(R) (40 g ai/ha) or Pydrin(R) (250 g ai/ha) was sprayed at 14 day intervals for leafroller and cowpea curculio control. Benlate(R) (550 g ai/ha) and Trigard (150 g ai/ha) were applied fortnightly for disease and leafminer control respectively. Mesurol(R) (200 g ai/ha) was applied as needed for snail and slug control. Treatments were replicated four times in a randomized complete block. Fresh pod weights were determined and yield loss computed from the undefoliated plot data at each growth stage. Yield data were analyzed by analysis of variance and regression utilizing the general linear models procedure of SAS (Ray, 1982).

Results

Defoliation had a significant effect on yield in both the greenhouse and the field, with F-values of 50.16 and 39.95 ($p < 0.001$) respectively (Table 2). There was no significant interaction between time of defoliation and defoliation levels under greenhouse conditions ($F = 0.79$). There was significant interaction between defoliation levels and time of defoliation in the field ($F = 3.83$). Analysis of variance on the effects of time of defoliation showed that there were significant differences between defoliation times ($F = 6.23$) in the field but not in the greenhouse.

Regression analysis of the relationship between yield in g/plot and defoliation level produced models of the form: $Y = a + bx$ where Y = log (yield), x = defoliation level and a = intercept. The quadratic model

($a + bx = cx^2$) resulted in higher coefficients of determination (R^2) (Table 3). Generally the fit of the quadratic models to the data was better than the linear model, although the increase in R^2 was generally less than 10%. Thus the predictive powers of the linear and quadratic models were more or less similar.

Defoliation did not reduce yield proportionally to its magnitude in both the greenhouse and the field (Tables 4, 5 and Figures 1, 3). Conversely, 25% defoliation resulted in yield increases at the primary leaf stage and full bloom in the field (Table 5). Defoliating plants at the first trifoliate leaf stage at 75% level increased yield by 4% under field conditions (Table 5). No yield increases due to defoliation at pod set in the greenhouse and at full bloom and pod set in the field were observed (Table 5). Total defoliation at pod set resulted in 74% yield loss in the greenhouse but losses of 95% and 92% yield loss at full bloom and pod set in the field.

Yield loss in the greenhouse ranged from 16% to 74% whereas in the field it ranged from -4% to 95% (Table 5). The least yield reduction in the greenhouse was observed at 50% defoliation when foliage was removed at pod set. There was, however, an increase in yield in the field when 75% of the foliage was removed at the first trifoliate leaf stage (Table 5).

Gross dollar values of 'Sprite' snap beans per hectare are shown in Tables 6 and 7 and Figures 5-10. These values were computed based the following price ranges $6.00 (low), $11.00 (medium), and $20.00 (high) for 13.62 kg of snap beans. Since the undefoliated control generally gave higher yields, dollar values obtained from it were higher. In the field, however, 75% defoliation at the first trifoliate leaf stage gave

TABLE 2. F-values from analysis of variance for the effects of defoliation, time (plant growth stage) and their interaction on snap bean yield.

	Field		Greenhouse	
Source	F	Prob. F	F	Prob. F
Defoliation	50.16	0.0001	39.95	0.0001
Growth Stage	6.23	0.0001	0.35	0.92
Defoliation X Growth Stage	3.83	0.001	1.04	0.79

TABLE 3. Regression equations for the relationship between yield and defoliation.

Plant Growth Stage	Greenhouse		Field	
		Linear		
Primary leaf	$y = 127.5 - 76x$	$R_2^2 = 0.48$	$y = 1525 - 805x$	$R_2^2 = 0.41$
	$(y = 2.11 - 0.39x)$[a]	$(R_2^2 = 0.49)$	$(y = 3.18 - 0.35x)$	$(R_2^2 = 0.34)$
First trifoliate leaf	$y = 108.1 - 4.64x$	$R_2^2 = 0.22$	$y = 1197 - 686x$	$R_2^2 = 0.16$
	$(y = 2.01 - 0.24x)$	$(R_2^2 = 0.22)$	$(y = 3.08 - 0.62x)$	$(R_2^2 = 0.25)$
Third trifoliate leaf	$y = 119.5 - 57.8x$	$R_2^2 = 0.35$	$y = 1420.4 - 922x$	$R_2^2 = 0.59$
	$(y = 2.08 - 0.31x)$	$(R_2^2 = 0.38)$	$(y = 3.2 - 0.55x)$	$(R_2^2 = 0.58)$
Flower bud formation	$y = 117 - 64.5x$	$R_2^2 = 0.42$	$y = 1286.5 - 863x$	$R_2^2 = 0.61$
	$(y = 2.07 - 0.36x)$	$(R_2^2 = 0.42)$	$(y = 3.14 - 0.54x)$	$(R_2^2 = 0.53)$
Full bloom	$y = 115.9 - 55.9x$	$R_2^2 = 0.30$	$y = 1266.2 - 999x$	$R_2^2 = 0.66$
	$(y = 2.05 - 0.31x)$	$(R_2^2 = 0.26)$	$(y = 3.36 - 1.47x)$	$(R_2^2 = 0.46)$
Pod set	$y = 125.2 - 76.2x$	$R_2^2 = 0.51$	$y = 1441 - 1390.3x$	$R_2^2 = 0.73$*
	$(y = 2.13 - 0.48x)$	$(R^2 = 0.51)$	$(y = 3.25 - 1.06x)$	$(R^2 = 0.81)$
		Quadratic		
Primary leaf	$y = 129.2 - 89.3x + 13.3x^2$	$R_2^2 = 0.48$	$y = 1446 - 173.6x - 631.4x^2$	$R_2^2 = 0.43$
	$(y = 2.07 - 0.14x - 0.25x^2)$	$(R_2^2 = 0.22)$	$(y = 3.15 - 0.08x - 0.26x^2)$	$(R_2^2 = 0.36)$
First trifoliate leaf	$y = 117 - 117.3x + 70.9x^2$	$R_2^2 = 0.27$	$y = 1005.6 + 845.4x - 1531.4x^2$	$R_2^2 = 0.24$
	$(y = 2.05 - 0.54x + 0.29x^2)$	$(R_2^2 = 0.25)$	$(y = 2.85 + 1.16x - 1.78x^2)$	$(R_2^2 = 0.43)$
Third trifoliate leaf	$y = 117.3 - 39.9x - 17.8x^2$	$R_2^2 = 0.34$	$y = 1269.8 + 283.7x - 1205.7x^2$	$R_2^2 = 0.68$*
	$(y = 2.05 - 0.01x - 0.23x^2)$	$(R_2^2 = 0.41)$	$(y = 3.07 + 0.52x - 1.07x^2)$	$(R_2^2 = 0.77)$
Flower bud formation	$y = 115.7 - 53.5x - 11x^2$	$R_2^2 = 0.43$	$y = 1284.7 - 848.7x - 14.3x^2$	$R_2^2 = 0.61$
	$(y = 2.03 - 0.06x - 0.3x^2)$	$(R_2^2 = 0.45)$	$(y = 3.1 - 0.17x - 0.37x^2)$	$(R_2^2 = 0.55)$
Full bloom	$y = 116.3 - 58.9x - 3x^2$	$R_2^2 = 0.29$	$y = 1129.8 + 91.1x - 1090.9x^2$	$R_2^2 = 0.73$*
	$(y = 2.03 - 0.16x - 0.15x^2)$	$(R_2^2 = 0.27)$	$(y = 2.94 + 1.86x - 3.32x^2)$	$(R_2^2 = 0.67)$
Pod set	$y = 114.4 + 10.7x - 86.9x^2$	$R_2^2 = 0.55$	$y = 1552 - 2282.6x + 892.3x^2$	$R_2^2 = 0.87$*
	$(y = 2.02 - 0.38x - 0.86x^2)$	$(R^2 = 0.65)$	$(y = 3.17 - 0.42x - 0.64x^2)$	$(R^2 = 0.84)$

[a] Figures in parentheses are y = log (yield); x = defoliation as proportion.

* R significant at 0.05.

TABLE 4. Effect of defoliation and defoliation time on snap bean yield (g/plot) in the greenhouse and field. Data are means of 4 replicates.

Defoliation level (%)	Snap bean yield (g/plot) by plant growth stage											
	Primary leaf		First trifoliate leaf		Third trifoliate leaf		Flower bud formation		Full bloom		Pod set	
	Greenhouse	Field	Greenhouse	Field	Greenhouse	Field	Greenhouse	Field	Greenhouse	Field	Greenhouse	Field
0	138	1361	122	1168	125	1293	128	1295	121	1138	122	1530
25	89	1403	84	835	97	1210	89	1113	94	1140	96	1085
50	92	1105	72	933	99	1140	89	673	79	690	102	495
75	84	1050	82	1215	81	820	78	620	93	813	84	457
100	46	613	65	120	54	335	46	350	52	52	32	125

TABLE 5. Relationship between defoliation, time and yield (loss (%)) of snap beans in the greenhouse and field.

Defoliation level (%)	Yield (% loss by plant growth stage											
	Primary leaf		First triboliate leaf		Third trifoliate leaf		Flower bud formation		Full bloom		Pod set	
	Greenhouse	Field	Greenhouse	Field	Greenhouse	Field	Greenhouse	Field	Greenhouse	Field	Greenhouse	Field
0	0	0	0	0	0	0	0	0	0	0	0	0
25	36	-3	31	29	22	16	30	14	22	0	21	29
50	34	19	41	20	21	12	30	48	35	39	16	68
75	39	23	32	-4	35	37	39	52	23	29	31	70
100	69	55	47	74	57	74	64	73	57	95	74	92

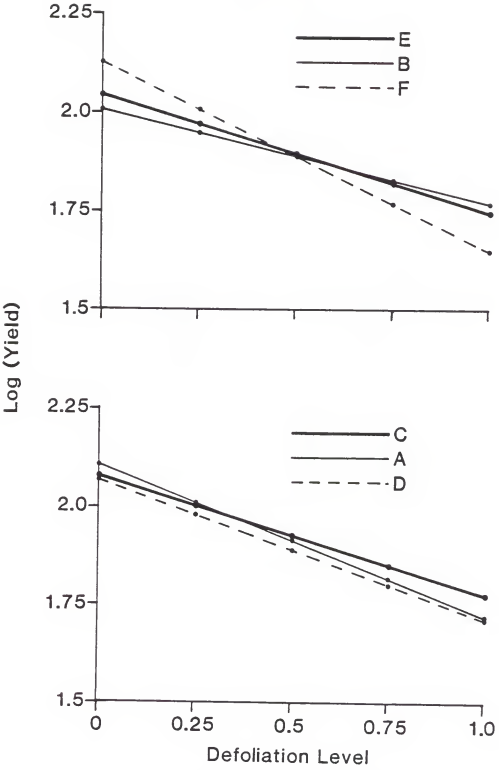


Figure 1. Effects of defoliation and time of defoliation on snap beans yield in the greenhouse (linear models).

Letters represent the plant growth stage defoliation occurred.
A = primary leaf, B = first trifoliate leaf, C = third trifoliate leaf,
D = flower bud formation, E = full bloom, and F = pod set.

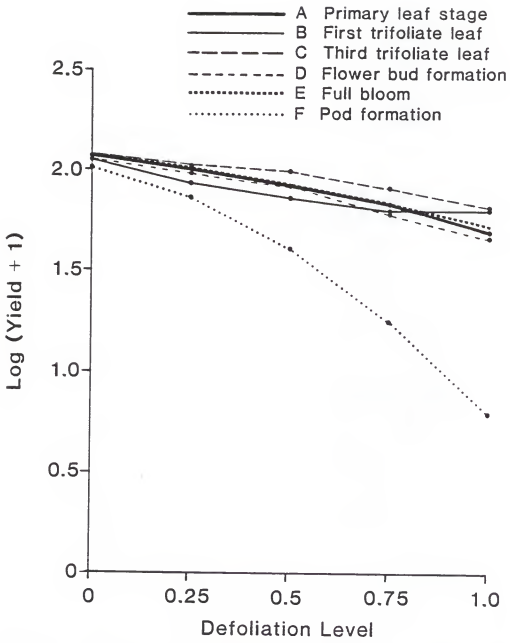


Figure 2. Effects of defoliation and time of defoliation on snap bean yield in the greenhouse (quadratic models).

Letters represent the plant growth stage defoliation occurred.

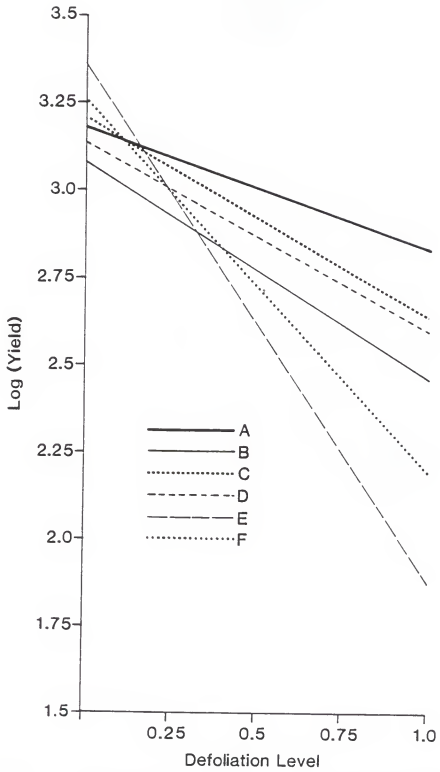


Figure 3. Effects of defoliation and time of defoliation on snap bean yield under field conditions (linear models).

Letters represent the plant growth stage defoliation occurred. A = primary leaf, B = first trifoliate leaf, C = thrid trifoliate leaf, D = flower bud formation, E = full bloom, and F = pod set.

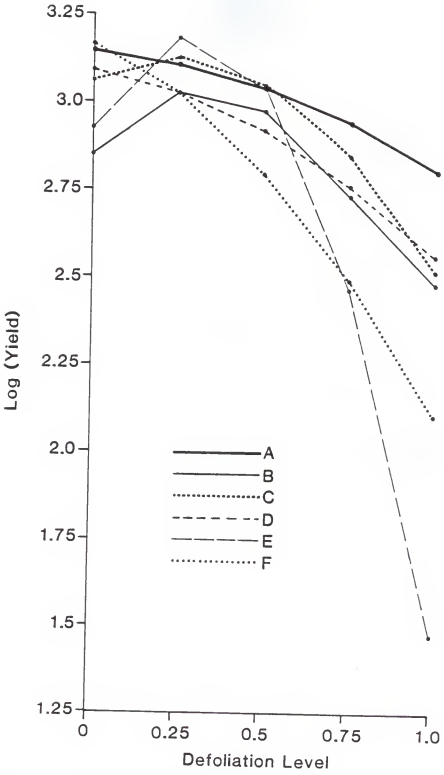


Figure 4. Effects of defoliation and time of defoliation on snap bean yield under field conditions (quadratic models).

Letters represent the plant growth stage defoliation occurred. A = primary leaf, B = first trifoliate leaf, C = third trifoliate leaf, D = flower bud formation, E = full bloom, and F = pod set.

TABLE 6. Effects of defoliation and defoliation time on gross dollar values per hectare of 'Sprite' snap beans grown in the greenhouse.

Defoliation level (%)	Price range[a]	Gross dollar values per hectare Time of defoliation (growth stage) Primary leaf	First trifoliate leaf	Third trifoliate leaf	Flower bud formation	Full bloom	Pod set
0	low	292	258	265	270	256	258
	medium	535	472	485	495	468	472
	high	974	859	885	900	852	859
25	low	187	178	207	189	199	204
	medium	343	326	379	347	365	373
	high	623	593	689	630	664	679
50	low	193	152	209	189	167	216
	medium	353	279	384	347	307	397
	high	643	507	698	630	558	772
75	low	178	175	172	165	197	178
	medium	327	321	316	302	361	326
	high	594	584	574	549	656	593
100	low	96	137	115	59	110	67
	medium	177	250	211	109	201	123
	high	332	455	384	198	366	223

[a] low = $7/13.62 kg; medium = $11.20/13.62 kg; and high = $20/13.62 kr of snap beans.

TABLE 7. Effects of defoliation and defoliation time (plant growth stage) on gross dollar values per hectare of Sprite snap beans grown in the field.

Defoliation level (%)	Price range[a]	Gross dollar values per hectare Time of defoliation (plant growth stage) Primary leaf	First trifoliate leaf	Third trifoliate leaf	Flower bud formation	Full bloom	Pod set
0	low	333	285	316	317	278	374
	medium	611	523	580	582	510	686
	high	1110	952	1054	1057	928	1248
25	low	343	176	266	273	279	266
	medium	629	323	487	500	511	487
	high	1143	588	886	909	930	886
50	low	270	228	278	165	170	157
	medium	495	419	510	302	311	288
	high	869	761	928	549	566	524
75	low	256	297	200	152	197	112
	medium	470	544	367	279	361	206
	high	855	990	666	507	656	374
100	low	150	74	82	86	110	30
	medium	275	136	151	157	201	55
	high	500	247	274	285	366	100

[a] Low = $7/13.62 kg; medium = $11.20/13.62 kg; and high = $20/13.62 kg of snap beans.

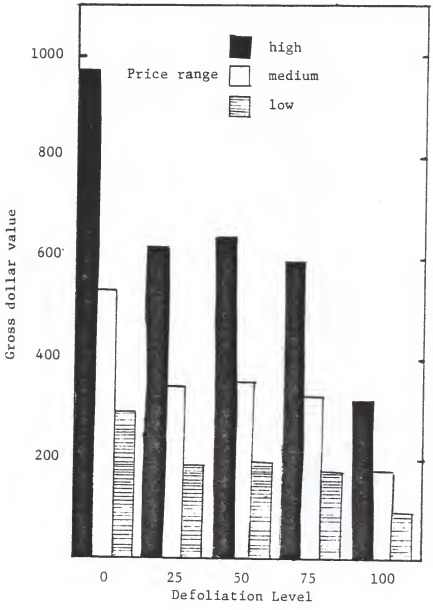


Figure 5. Influence of defoliation on gross dollar value per hectare of 'Sprite' snap beans defoliated at the primary leaf stage in the greenhouse.

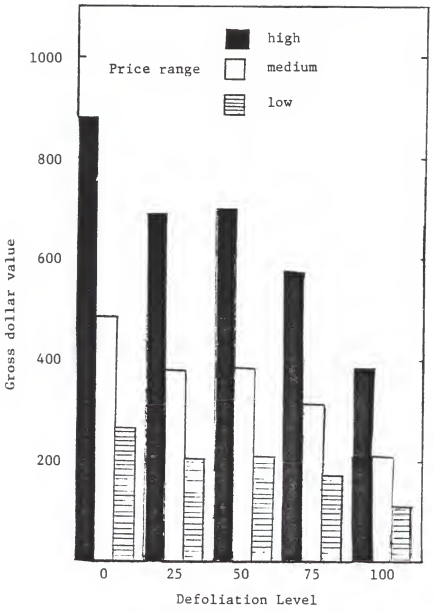


Figure 6. Influence of defoliation on gross dollar value per hectare of 'Sprite' snap beans defoliated at the third trifoliate leaf stage in the greenhouse.

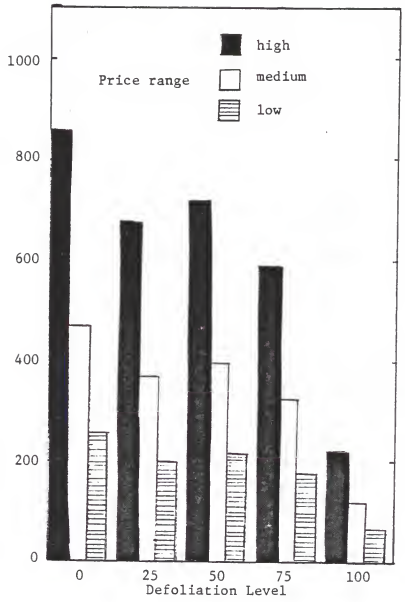


Figure 7. Influence of defoliation on gross dollar value per hectare of 'Sprite' snap beans defoliated at pod set in the greenhouse.

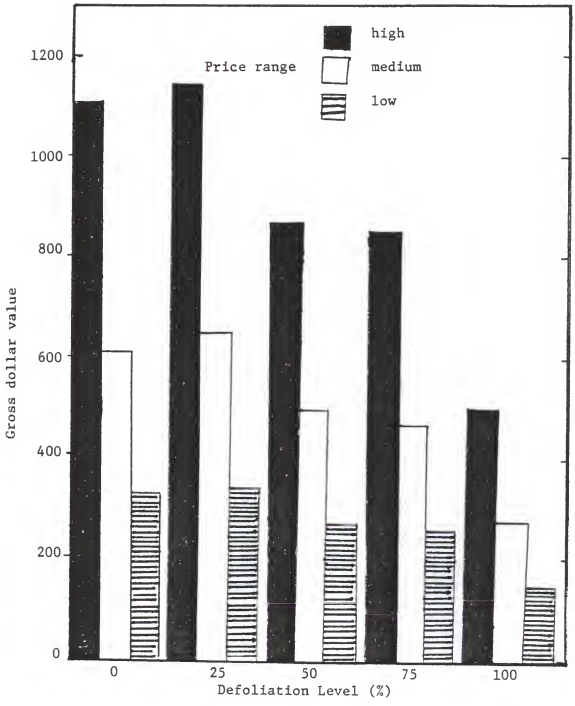


Figure 8. Influence of defoliation on gross values per hectare of 'Sprite' snap beans defoliated at the primary leaf stage in the field.

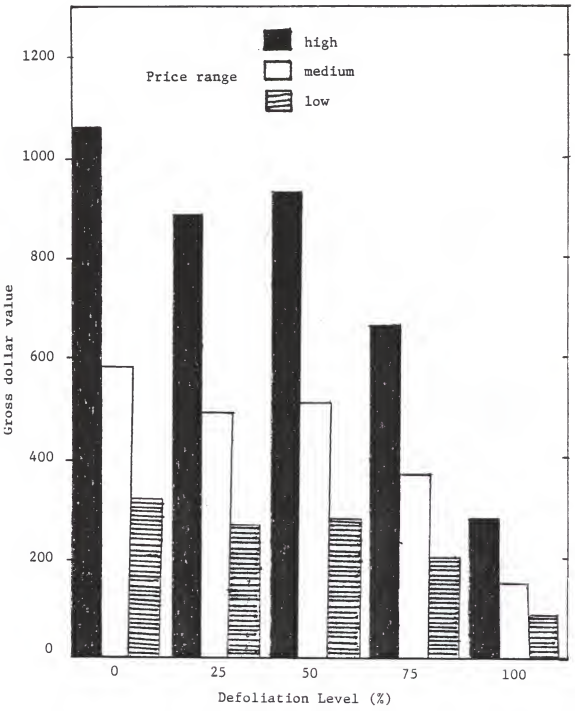


Figure 9. Influence of defoliation on gross dollar values per hectare of 'Sprite' snap beans defoliated at the third trifoliate leaf stage in the field.

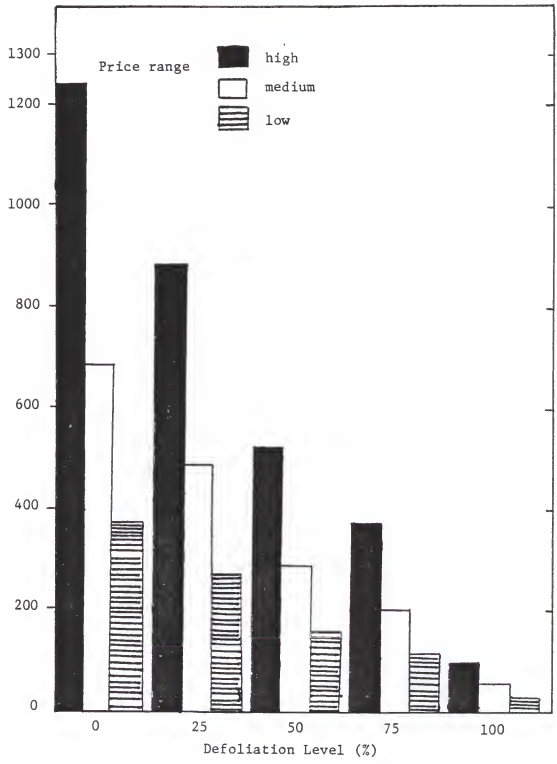


Figure 10.. Influence of defoliation on gross dollar values per hectare of 'Sprite' snap beans defoliated at pod set in the field.

a higher dollar value than the undefoliated control. Removal of 25% of the foliage at the primary leaf stage and full bloom also resulted in slightly higher dollar values than the undefoliated control. Generally, increased defoliation resulted in lower dollar values per hectare.

Discussion

Total defoliation when only primary leaves were present resulted in 69% and 55% yield loss in the greenhouse and field respectively. This level of defoliation resulted in 57%, 95%, 74% and 92% yield reduction in the greenhouse and field when plants were defoliated at full bloom and pod set, respectively. The lower yield reduction in the greenhouse may be due to the better controlled environmental conditions. The only growth stage at which total defoliation resulted in less yield loss in the field was at the primary leaf stage. This may have been due to better recovery of plants from the total defoliation in the field.

Removing 25% of the foliage resulted in yield loss of at least 20% in the greenhouse at all growth stages. This is a substantial loss in terms of dollar values. Thus, it appears that the economic threshold under greenhouse conditions was between 0 and 25% defoliation. In the field the economic threshold level varied with the plant growth stage (Table 4). The increase in yield in the field may have been due to compensatory reactions of the plant. The compensation may have resulted from increased photosynthesis due to increased exposure of the remaining photosynthetically active foliage to light. The same may apply to the increase in yield of plants with 75% defoliation at the first trifoliate leaf stage. Reducing foliage may have increased air circulation among

plants which, indirectly, may have increased carbon dioxide uptake and hence photosynthesis. Increased air circulation may also have resulted in reduced leaf surface humidity which may have reduced subtle fungal diseases from being established on the crop.

Removing all leaves from plants at pod set and full bloom resulted in substantial loss under both conditions probably because at these stages the developing pods were deprived of photosynthates normally manufactured in these leaves. The pods which developed probably utilized reserved photosynthates initially and thereafter photosynthates which were produced from the few leaves which were formed after defoliation. At the primary leaf stage, total defoliation slowed down the growth rate of the plants. Under greenhouse conditions, recovery may have been slow and plants may have been etiolated due to insufficient light, hence the higher yield loss. In the field, total defoliation at the first trifoliate leaf stage through flower bud formation resulted in 74 and 73% yield loss. This loss in yield is essentially similar in magnitude indicating that the sensitivity of plants at these stages was more or less the same.

Results obtained in these experiments seem to show yield increases due to defoliation. This may have been due to chance effects resulting from many factors including plant characteristics and the environment. Generally, data show tendency to lower production and hence dollar values with increased defoliation. The decrease in production due to defoliation may have been to enhancement of pathogen entry through wounds made during manual defoliation. Defoliating with scissors also led to water loss through direct evaporation. Defoliation also reduced the leaf area for photosynthesis. One can only suspect that the

inconsistency was due to the imprecise nature of manual defoliation in simulating insect damage. It is possible that removing whole fractions of leaf surfaces had a different effect on plants from damage done by leaf feeding pests which usually occurs at random.

Although the influence of defoliation on yield was not consistent at all plant growth stages, plants showed more sensitivity at full bloom and pod set. This was an indication that leaf damaging pests should be managed before these plant growth stages. If left unchecked and if plants become heavily defoliated, substantial loss in yield would be expected. Insecticides are generally applied as soon as insect pest infestations are detected. Insecticide application normally starts before the pest populations exceed threshold levels. Disease control chemicals are primarily preventatives applied well before the diseases are observed. Since pesticide sprays against diseases and insect pests were the same at all defoliation levels and plant growth stages, the grower would incur loss in gross dollar values proportional to loss in yield. In this study the threshold level for defoliation was below 25%. At all plant growth stages.

CHAPTER IV
THE EFFECT OF ROOT-KNOT NEMATODES AND DEFOLIATION ON SNAP BEANS

Introduction

Root-knot nematodes, especially Meloidogyne incognita (Kofoid and White) Chitwood, are a serious threat to bean (Phaseolus vulgaris L.) production in many bean-producing areas of the world (Agudelo, 1980; Allen, 1983; Ngundo, 1977; Sharma and Guazelli, 1982; Singh et al. 1981a). M. incognita has been reported to cause extensive root-galling on bean plants, which interferes with nitrogen fixation by Rhizobium spp. as well as nutrient uptake by the root system. Moreover, M. incognita has been reported to increase the severity of other pathogens through predisposition of host plant root tissues (Carter, 1975a,b; Golden and Van Gundy, 1975; Powell, 1971; Powell and Nusbaum, 1960; Porter and Powell, 1967; Sasser et al., 1955). Thus, yield loss caused by M. incognita and related species is not always a unitary effect, but often a result of interaction of these nematodes with other plant-parasitic organisms.

Yield losses of 50 to 90% in field beans have been reported due to severe root-knot nematode infections (Agudelo, 1980; Freire and Ferraz, 1977; Ngundo, 1977; Varon and Galvez, 1974). Yield decreases caused by M. incognita are also well known in other crops (Allen, 1983; Lamberti, 1979). Crop yield is usually expected to be inversely related to nematode counts (Sasser et al., 1968). In view of this theory, Barker

et al. (1976), Di Vito et al. (1981), and Di Vito and Ekanayake (1983) reported the relationship between initial M. incognita densities and plant growth or yield of tomato and sugar beet. Barker et al. (1976) showed that M. incognita suppressed yields of tomato in North Carolina by up to 85% in the coastal plains and 20-30% in mountain locations. Meloidogyne incognita has been observed to cause yield losses of 30-60% and up to 15% in eggplant and pepper (Capsicum frutescens L.) respectively (Lamberti, 1975). Yield losses due to M. incognita infections have been reported on okra (Hibiscus esculentus L.), sweet potato (Ipomoea batatas (L.) Lam.), celery (Apium graveolens L.), and carrot (Daucus carota L.) (Lamberti, 1971).

Root-knot nematodes rarely occur alone on any crop (Powell, 1971). Thus, nematodes may occur together with other plant pests and diseases. McSorley and Waddill (1982) reported yield loss partitioning on yellow squash (Cucurbita pepo L.) into nematode and insect components by using multiple regression. The partitioning of yield loss was facilitated by the use of selective pesticides. Consequently, McSorley and Waddill (1982) suggested that it may be imperative to separate pests into nematode and insect components when complexes of several pests were present. This separation of yield loss components would be facilitated by monitoring field pest populations during the growing season, at specific intervals, to detect population changes (McSorley and Waddill, 1982).

Beans are susceptible to defoliation by insects, adverse environmental conditions, diseases, and mechanical injury (Agudelo, 1980). Hence an understanding of the relationship between crop yield and pest infestations is essential for the development of sound pest management

strategies. One way of elucidating this relationship has been manual defoliation of plants to simulate pest damage (Douglas et al., 1981; Galvez et al., 1977; Greene and Minnick, 1967; Hohmann and De Carvalho, 1983; Kalton et al., 1945; Keularts, 1980; Wit, 1983, Wolk et al., 1983). Hohmann and De Carvalho (1983) reported that leaf area reduction of 25, 50, 75, and 100% on the bean cultivar Carioca reduced yield by 11, 20, 20, and 70% respectively when defoliation was done at the pod formation stage. At the same percentage of leaf area reduction, defoliation at initiation of flowering decreased yield by 18, 12, 19 and 55% respectively. Greene and Minnick (1967) indicated that yield reduction in 'Harvester' snap beans due to leaf removal began somewhere between 33% and 50% defoliation when plants were defoliated in the bloom or pre-bloom stages. Working on inderterminate snap beans, Waddill et al. (1984) noted that the removal of both primary leaves when only primary leaves were present resulted in yield reduction of up to 65%. Vieira (1981) reported that 66% defoliation of an indeterminate bean cultivar at the flowering and pod formation stages was detrimental to yield. Galvez et al. (1977) observed that total defoliation at the formation of the first trifoliate leaves reduced yields of the bean cultivars ICA-Guali and Porrillo-Sentetico by 34% and 49% respectively. These observations indicate that the magnitude of yield loss due to defoliation depends not only on the severity of defoliation but also on the growth stage (time) the defoliation takes place and the cultivar of beans grown. Thus, manual defoliation provides a useful estimate of the host-pest relationship despite its imprecision in simulating pest damage (Ruesink and Kogan, 1975).

This study was conducted to investigate the relationship between M. incognita population levels, manual defoliation, and their interaction to snap bean yield.

Materials and Methods

Two studies were conducted in a greenhouse at the Tropical Research and Education Center in Homestead, Dade County, Florida, in the summer and fall 1984. One experiment was an inoculation experiment with M. incognita designed to determine the effect of this pest alone on yield, and the other experiment examined the simultaneous effect of M. incognita inoculation and manual defoliation. The first experiment (M. incognita alone) was designed as a randomized complete-block replicated four times, involving six different nematode population levels. The other experiment was a 5x4x6 factorial replicated four times and included the following treatments: 5 defoliation levels of 0, 25, 50, 75, and 100%; 4 nematode population levels of 0, 1,000, 10,000, and 100,000 eggs and juveniles per pot; and defoliation at the following 6 plant growth stages: primary leaf, first trifoliate leaf, third trifoliate leaf, flower bud formation, full bloom, and pod set.

Preparations for both experiments were made by covering Rockdale soil (3030 L) with a polyethylene sheet and fumigating it with Dowfume$^{(R)}$ MC2 (681g). Two -gallon, side-drain, plastic pots were filled with 6.4 L soil and placed on corrugated greenhouse benches 0.91 m high. Fertilizer (8:16:16) was applied before planting at 3 g/pot (448 kg/ha). Plants were top-dressed with the same fertilizer at 1.5 g/pot four weeks after germination. The M. incognita inoculation test

was planted on 25 June 1984 and harvested on 28 August 1984. The M. incognita x defoliation study was planted on 24 October 1984 and harvested on 26 December 1984 to 3 January 1985. In each experiment, a plot comprised of 2 pots, each containing 3 plants. Irrigation was provided by an automatic time-controlled, overhead, water-mist-producing system twice a day.

The Meloidogyne incognita population used in each experiment was originally obtained from Hausa potato (Coleus parviflorous Benth.) and was maintained on greenhouse-grown tomato (Lycopersicon esculentum Mill.) 'FloraDade' plants. Meloidogyne incognita eggs were extracted by the sodium hypochlorite (NaOCl) method of Hussey and Barker (1973). A 0.525% NaOCl solution was made from Thrift King(R) commercial bleach (5.25% NaOCl) by dilution with cold tap water (25°C). Tomato roots were thoroughly washed of soil with running tap water. The clean roots were cut into 2-3 cm long pieces and 120 g of the cut root material was manually shaken in 200 ml of the NaOCl solution for 3.5 minutes. The shaken material was serially filtered through 100-mesh, 230-mesh and 500-mesh sieves. The number of eggs and juveniles of M. incognita per ml was determined by counting in a watch glass under a dissecting microscope (20X). Appropriate dilutions of the nematode eggs and juveniles were made according to the population levels used.

Plants were inoculated 10 days after planting by drenching their bases with the nematode egg and juvenile suspension. The nematode population levels of 0, 10, 100, 1,000, 10,000, and 100,000 per pot were equivalent to 0, 0.16, 1.6, 15.6, 156 and 1562 eggs and juveniles per 100 ml soil, respectively.

In the experiment involving simultaneous nematode inoculation and defoliation, plants were defoliated manually with a pair of scissors. The correct number or proportion of leaves to be removed was determined by leaf counts at each plant growth stage. To eliminate additional uncontrolled defoliation, plants were sprayed with Ambush(R) (40 g ai/ha) for bean leaf roller (Urbanus proteus L.) and cowpea curculio (Chalcodermus aeneus Boh.) control; Trigard(R) (150 g ai/ha) for leafminer (Liriomyza spp.) control, and Benlate(R) (550 g ai/ha) for disease control. These pesticides applied at 14-day intervals. Yield was taken from all six plants. Pods less than 7 cm in length and diseased or damaged ones were discarded.

Yield data were subjected to analysis of variance and regression analysis using the general linear models procedure of SAS (Ray, 1982). Nematode inoculation data were also analyzed using Seinhorst's models (Ekanayake and Di Vito, 1984; Ferris, 1984; Ferris et al., 1981; Seinhorst, 1965).

Results

Meloidogyne incognita alone

Analysis of variance on the effect of Meloidogyne incognita population levels on yield showed a significant relationship with $F = 26.2^{***}$ (Table 8). Regression analysis of the data produced models of the form $Y = a + bx$ or $Y = a + bx + cx^2$ where Y = yield (g/plot) or log (yield), x = log (M. incognita population + 1) (Table 9). Quadratic models consistently gave somewhat higher coefficients of determination (R^2) values. The predictive ability of the quadratic models was, however,

TABLE 8. Effect of M. incognita on snap bean yield. Data are means of 4 replicates.

No. of M. incognita/Pot	Log (M. incognita Population + 1)	Yield (g/plot)[a]	Yield loss (%)
0	0	128	0
10	1.0	103	19
100	2.0	79	38
1000	3.0	70	45
10000	4.0	68	47
100000	5.0	53	59

[a] Data rounded off to the nearest whole number.

F = 26.2*** for M. incognita populations.

not greatly superior to that of linear models (Figure 11). Yield reduction was initiated even by the lowest nematode population.

The data did not fit the Seinhorst (1965) model, which is of the form $Y = m + (1-m)\ Z^{[P-T]}$, where Y = ratio between yield at nematode population level p and in the absence of nematodes, m = minimum yield at very high nematode population levels, T = tolerance limit (the nematode population level below which yield reduction does not occur), and Z = the proportion of the plant undamaged in the presence of parasitism or infection by one nematode (Ferris et al., 1981; Ferris, 1984; Seinhorst, 1965 and 1972). The Seinhorst model produced a R^2-value of only 0.00661, an indication that the data had a poor fit to the model. Data were, moreover, linear in trend (Figure 11).

Table 10 shows the gross dollar values per hectare of snap beans. These values were computed from the price ranges of $6.00 (low), $11.00 (medium), and $20.00 (high) per bushel (13.62 kg) of beans. These were derived from gross yield and prices given above. They are what the grower would get without subtracting the cost of production which includes labor, pesticides, rent, farm machinery depreciation, and/or interest in loans. The nematode-free plots produced at least twice as much money as plots with 10,000 or 100,000 eggs and juveniles of M. incognita per pot which were equivalent to 156 and 1562 eggs and juveniles per 100 ml soil. Nematode-free plants produced $404, $740 and $1345 per hectare at the low, medium, and high prices respectively compared to $326, $598, and $1087 when pots were inoculated with 10 eggs and juveniles (= 0.16/100 ml soil). This level of M. incognita population resulted in a 19% loss in gross dollar value per hectare ($77, $142, and $258 loss at low, medium and high prices, respectively).

Meloidogyne incognita inoculation x defoliation

Analysis of variance on the relationship between yield and M. incognita defoliation, and their interaction showed that there was no significant interaction between defoliation and M. incognita at all plant growth stages during which defoliation was done (Table 11). There were, however, significant differences among M. incognita population levels, and there were also significant differences in yield with defoliation level at all plant growth stages (Table 11). Regression analysis produced models of the form $Y = a + bx + cn$, where Y = yield (g/plot, x = defoliation level (as a fraction), and n = log (M. incognita population + 1) (Table 12). Quadratic models of the form $Y = a + b_1x + b_2x^2 + c_1n + C_2n^2$ gave somewhat higher coefficients of determination (R^2) (Table 12), but are more difficult to visualize graphically. In general, yield was reduced much faster by nematodes than when defoliation was held constant then when nematode populations were held constant and defoliation levels were changed (Figure 13).

Table 13 shows data on the effects of defoliation, and M. incognita, when they occurred simultaneously, on snap beans. The lowest yields were obtained when 100% of the leaf area was removed at the first trifoliate leaf stage or at flower bud formation and plants were inoculated with 100,000 eggs and juveniles/pot (Table 13). Total defoliation at any time from the third trifoliate leaf stage through to pod set reduced yield by at least 93% in the presence of an initial nematode population density of 100,000 eggs and juveniles/pot. There was, generally, no apparent synergistic effect when defoliation and M. incognita occurred simultaneously (Tables 13 and 14), which was evidenced by the lack of significance for the interaction term in the analysis of variance.

TABLE 9. Regression equations for the relationship between M. incognita population levels (x) and yield component (y).

Yield component	Model	
	Linear	Quadratic
Yield	$Y = 118.6 - 13.98x$ $R^2 = 0.81^*$	$Y = 126.7 - 26x + 2.4x^2$ $R^2 = 0.87^*$
Log (Yield)	$Y = 2.1 - 0.1x$ $R^2 = 0.84^{**}$	$Y = 2.1 - 0.1x + 0.005x^2$ $R^2 = 0.85^*$

* R significant at 0.05

** R significant at 0.01

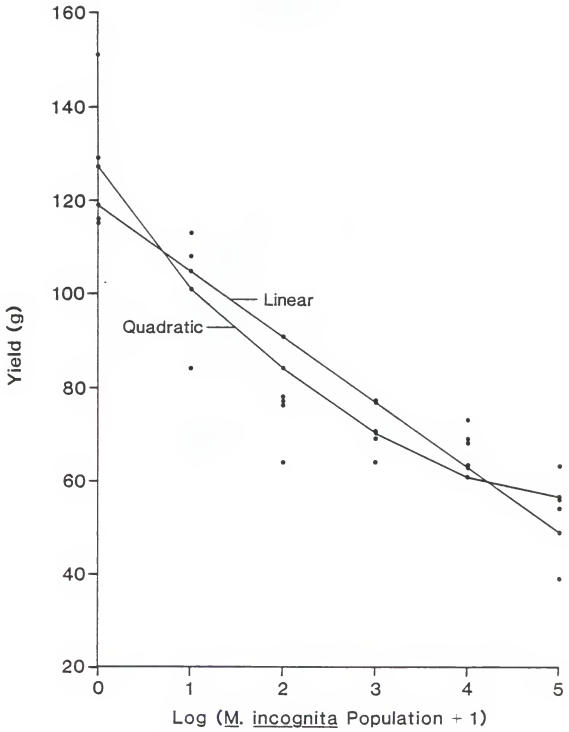


Figure 11. Effects of M. incognita on snap bean yield.

TABLE 10. The influence of M. incognita on the gross dollar value per hectare of snap beans grown in a greenhouse.

Log (M. incognita population +1)	Price range[a]	Gross dollar values per hectare
0	low	404
	medium	740
	high	1345
1.04	low	326
	medium	598
	high	1087
2.0	low	248
	medium	456
	high	828
3.0	low	222
	medium	407
	high	740
4.0	low	215
	medium	395
	high	718
5.0	low	167
	medium	307
	high	558

[a] Low = $6; medium = $11; high = $20/13.62 kg of snap beans.

TABLE 11. F-values and probability levels from analysis of variance for the effects of defoliation and M. incognita and their interaction on snap bean yield.

Source	Plant Growth Stage											
	Primary leaf		First trifoliate leaf		Third trifoliate leaf		Flower bud formation		Full bloom		Pod set	
	F	Prob. F	F	Prob. F	F	Prob. F	F	Prob. F	F	Prob. F	F	Prob. F
Defoliation	17.67	0.0001	67.84	0.0001	66	0.0001	41.6	0.0001	100	0.0001	140	0.0001
Log (M. incognita population +1)	23	0.0001	23.5	0.0001	35	0.0001	38	0.0001	28.95	0.0001	56.15	0.0001
Defoliation x Log (M. incognita population +1)	0.56	0.8871	0.54	0.8395	1.5	0.0962	0.72	0.7396	1	0.4601	0.38	0.9575

TABLE 12. Regressions equations for the relationship between M. incognita defoliation and yield.

Growth stage plants were defoliated	Regression equation[a]
Linear equations	
Primary leaf stage	$Y = 259-22.12N-25.1x$ ($R^2 = 0.62$**)
First trifoliate leaf stage	$Y = 256.51-21.04N-32.3x$ ($R^2 = 0.59$**)
Third trifoliate leaf stage	$Y = 267.04-21.51N-33x$ ($R^2 = 0.72$**)
Flower bud formation stage	$Y = 258.15-24.17N-30.19x$ ($R^2 = 0.64$**)
Full bloom stage	$Y = 237.26-17.75N-31.34x$ ($R^2 = 0.58$**)
Pod set stage	$Y = 25176-21.44N-29.77x$ ($R^2 = 0.58$**)
Quadratic equations	
Primary leaf stage	$Y = 241.97 + 2.07N-5.13N^2-7.9x-4.3x^2$ ($R^2 = 0.66$**)
First trifoliate leaf stage	$Y = 227.11 + 5.39N-5.61N^2+8.09x-10.1x^2$ ($R^2 = 0.67$**)
Third trifoliate leaf stage	$Y = 239.73 - 2.42N-4.03N^2+7.42x-10.1x^2$ ($R^2 = 0.79$**)
Flower bud formation stage	$Y = 243.49 - 10.31N-2.94N^2-10.53x-4.94x^2$ ($R^2 = 0.66$**)
Full bloom stage	$Y = 204.27-0.57-3.64N^2+22.68x-13.5x^2$ ($R^2 = 0.79$**)
Pod set stage	$Y = 218.66-0.24N-4.5N^2+21.65x-12.86x^2$ ($R^2 = 6.68$**)

[a] Y = Yield (g/plot).
N = Log (M. incognita population + 1).
X = Defoliation level (fraction).
** = R significant at 0.01.

TABLE 13. Effects of M. incognita and defoliation on snap bean yield (g/plot). Data are means of 4 replicates.

Defoliation level	Log (M. incognita population +1)	Snap bean yield (g/plot) by plant growth stage					
		Primary leaf	First trifoliate leaf	Third trifoliate leaf	Flower bud formation	Full bloom	Pod set
0	0	261	278	274	288	238	247
0	3.0	219	247	230	207	188	193
0	4.0	173	163	150	155	122	143
0	5.0	112	96	102	87	87	89
0.25	0	213	166	225	215	224	220
0.25	3.0	186	138	173	148	182	175
0.25	4.0	139	118	141	129	141	135
0.25	5.0	84	60	121	95	110	106
0.50	0	217	235	204	196	187	203
0.50	3.0	172	183	170	145	157	154
0.50	4.0	147	140	153	94	126	111
0.50	5.0	87	107	114	71	98	74
0.75	0	185	188	193	180	145	216
0.75	3.0	145	156	156	144	135	170
0.75	4.0	106	114	87	95	109	136
0.75	5.0	92	80	66	80	69	66
1.0	0	126	46	80	83	48	38
1.0	3.0	87	30	50	50	28	35
1.0	4.0	58	21	29	27	17	23
1.0	5.0	41	12	19	16	17	15

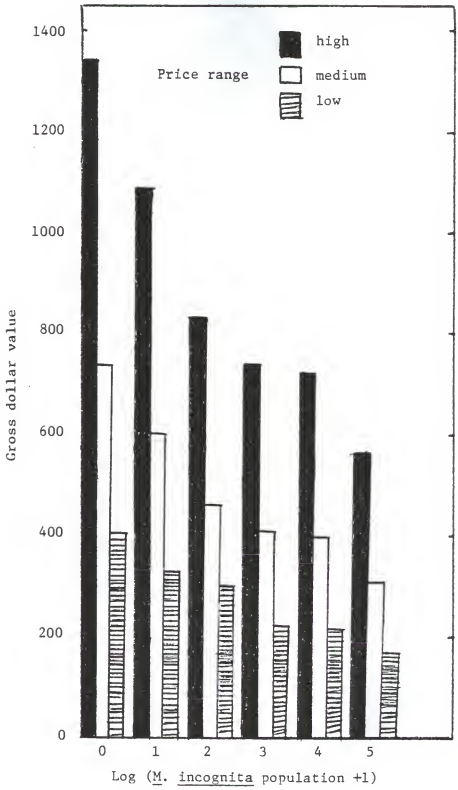


Figure 12. The influence of M incognita on gross dollar value per hectare of 'Sprite' snap beans.

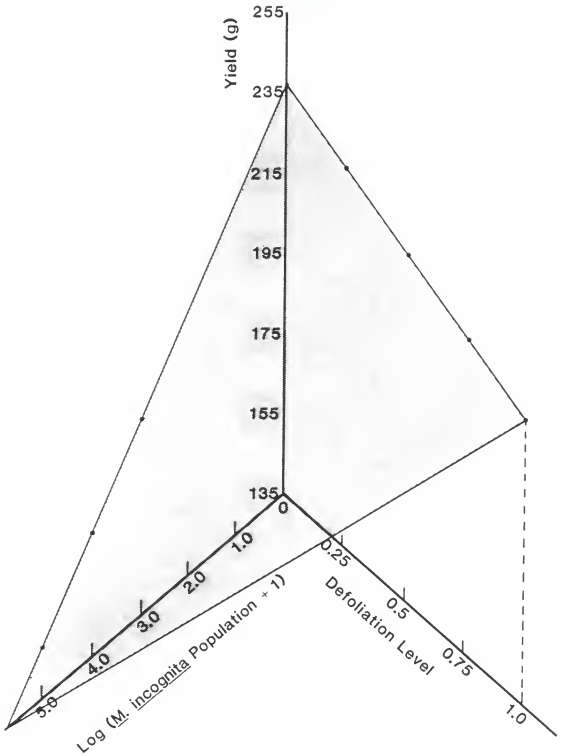


Figure 13: Effects of defoliation and M. incognita on snap bean yield.

TABLE 14. The influence of *M. incognita* and defoliation on snap bean yield loss (%).

Defoliation level	Log (*M. incognita* population +1)	Yield loss (%) by plant growth stage					
		Primary leaf	First trifoliate leaf	Third trifoliate leaf	Flower bud formation	Full bloom	Pod set
0	0	0	0	0	0	0	0
0	3.0	16	11	16	28	21	22
0	4.0	34	41	45	46	49	42
0	5.0	57	65	63	70	62	60
0.25	0	19	40	18	25	6	11
0.25	3.0	29	50	37	49	24	29
0.25	4.0	47	58	49	55	41	45
0.25	5.0	68	78	56	67	53	57
0.50	0	17	15	26	32	21	18
0.50	3.0	34	34	38	50	34	38
0.50	4.0	44	50	44	67	47	55
0.50	5.0	67	62	58	75	59	70
0.75	0	29	32	30	38	39	12
0.75	3.0	45	44	43	50	43	31
0.75	4.0	60	59	68	67	54	45
0.75	5.0	65	71	76	72	71	73
1.0	0	52	83	71	71	80	84
1.0	3.0	67	89	82	83	88	86
1.0	4.0	78	93	90	91	93	91
1.0	5.0	84	96	93	94	93	94

TABLE 15. Effects of *M. incognita* and defoliation on the gross dollar values per hectare of snap beans.

Defoliation level	Log (*M. incognita* population +1)	Price range	Gross dollar values by plant growth stage					
			Primary leaf	First trifoliate leaf	Third trifoliate leaf	Flower bud formation	Full bloom	Pod set
0	0	low	830	882	870	916	755	783
		medium	1522	1617	1595	1679	1384	1435
		high	2768	2940	2900	3053	2517	2609
0	3.0	low	697	785	731	659	596	611
		medium	1278	1439	1340	1209	1094	1119
		high	2325	2617	2436	2198	1988	2035
0	4.0	low	548	520	478	495	385	454
		medium	1005	954	877	907	706	833
		high	1827	1734	1595	1649	1283	1514
0	5.0	low	357	309	322	275	284	313
		medium	655	566	590	504	521	574
		high	1191	1029	1073	916	956	1044
0.25	0	low	673	529	714	687	710	697
		medium	1233	970	1308	1259	1301	1278
		high	2242	1764	2379	2289	2366	2323
0.25	3.0	low	589	441	548	467	574	556
		medium	1081	808	1005	856	1052	1019
		high	1965	1470	1827	1557	1913	1853
0.25	4.0	low	440	370	444	412	445	430
		medium	807	679	813	756	817	789
		high	1467	1235	1479	1374	1485	1435

TABLE 15. continued

Defoliation level	Log (*M. incognita* population +1)	Price range	Gross dollar values by plant growth stage: Primary leaf	First trifoliate leaf	Third trifoliate leaf	Flower bud formation	Full bloom	Pod set
0.25	5.0	low	266	194	383	302	355	337
		medium	487	355	702	554	651	617
		high	885	641	1276	1007	1184	1122
0.5	0	low	689	750	644	586	596	642
		medium	1264	1375	1181	1075	1094	1177
		high	2298	2499	2147	1954	1988	2140
0.5	3.0	low	548	582	540	458	586	485
		medium	1005	1067	989	840	1075	890
		high	1827	1940	1799	1527	1954	1618
0.5	4.0	low	465	441	482	302	398	352
		medium	853	808	893	554	730	646
		high	1551	1470	1624	1003	1327	1175
0.5	5.0	low	274	335	366	229	310	235
		medium	502	615	670	420	568	430
		high	912	1118	1219	764	1032	783
0.75	0	low	589	600	522	568	461	689
		medium	1081	1099	957	1041	845	1263
		high	1965	1999	1740	1893	1536	2297
0.75	3.0	low	457	494	496	458	430	540
		medium	838	905	909	840	789	990
		high	1523	1646	1653	1527	1435	1800

TABLE 15. continued.

Defoliation level	Log (*M. incognita* population +1)	Price range	Gross dollar values by plant growth stage					
			Primary leaf	First trifoliate leaf	Third trifoliate leaf	Flower bud formation	Full bloom	Pod set
0.75	4.0	low	332	362	278	302	347	430
		medium	609	663	510	554	636	789
		high	1107	1206	928	1007	1157	1435
0.75	5.0	low	291	256	209	256	219	133
		medium	533	469	383	470	401	244
		high	969	853	696	855	730	443
1.0	0	low	399	150	252	266	151	125
		medium	731	275	463	488	277	229
		high	1329	499	841	885	504	417
1.0	3.0	low	274	97	153	155	91	110
		medium	502	178	288	285	167	201
		high	912	323	523	518	303	366
1.0	4.0	low	183	62	87	82	53	70
		medium	335	113	160	151	97	129
		high	609	206	291	275	176	235
1.0	5.0	low	133	35	61	55	53	47
		medium	244	65	111	101	97	86
		high	443	117	203	184	176	157

Gross dollar values are shown in Table 15 and Figure 12. These values were computed from gross yield per hectare based on the following price ranges $6.00 (low), $11.00 (medium), and $20.00 (high). Hence these are gross values without deducting production costs. The gross dollar values (Table 15) show that there was a wide range at each plant growth stage. Generally highest dollar values were obtained when the plants were nematode-free and when no defoliation occurred. The combination of nematodes and defoliation had inconsistent effects on gross dollar values. If defoliation were held constant at any level, gross dollar values decreased as the nematode populations were increased (Table 15). If nematode populations were held constant the decrease in gross dollar values was not always consistent with the levels of defoliation. This is shown clearly at the primary leaf, first trifoliate leaf, full bloom and pod set stages of plant growth. At these stages some lower levels of defoliation have smaller dollar values than higher defoliation levels. As expected, loss in dollar values was similar to yield loss since the former was obtained from yield (g/plot), but varies greatly depending on the current market price which can fluctuate widely.

Discussion

There were significant differences in yield when M. incognita inoculated to snap beans. In this test 10 eggs and juveniles/pot reduced yield by at least 19%. In the defoliation - M. incognita interaction test, 1,000 eggs and juveniles/pot depressed yield by only 11-28% when plants were not defoliated (Table 14). Similar trends

occurred when 10,000 or 100,000 eggs and juveniles/pot were used. This discrepancy in M. incognita effects on snap bean yield may be due to the difference in the seasons in which the two trials were conducted. Tyler (1933) reported that at temperatures ranging from 27.5°C to 30°C, females of Meloidogyne spp. developed from infective juveniles to the egg-laying stage in 17 days; at 24.5°C in 21 to 30 days; at 20°C, in 31 days; at 15.4°C in 57 days; and at temperatures above 33.5%C or below 15.4°C, females failed to reach maturity on tomato plants. Decker and Casamayor-Garcia (1966) stated that one generation of M. incognita developed on lettuce within 26 days at a mean temperature of 23.3°C. They further stated that from the time of larval invasion up to the commencement of egg-laying required at least 19 days. Lamberti (1979) observed that M. incognita rarely started to invade root tissues when the soil temperature was below 18°C. Since the two tests being discussed here were conducted at different times of the year, it is likely that in the summer test soil temperatures were generally higher than in the fall trial. Thus, the life cycle of the nematode may have been completed in a shorter period of time in the summer than in the fall. Consequently, more M. incognita generations (at least 3) may have been completed during this season. There is also the possibility that the quality of the inoculum was different in the two tests since the source of the eggs and juveniles for the fail test was also exposed to relatively lower temperatures than the summer inoculum. The verification of this phenomenon can only be obtained by conducting further tests. The higher yields in the fall test may also be due to the fact that the nematodes were not able to invade the root tissues of the plants as fast as they could under optimum summer conditions. During

these tests soil temperatures were not taken which in fact precludes the comparison of edaphic temperatures during the time the two tests were conducted. Soil temperatures are generally warmer when air temperatures are higher.

The results obtained in the test where M. incognita was used alone indicate that the threshold population level was between 0 and 10 eggs and juveniles/pot whereas in the fall experiment the threshold population level was between 0 and 1,000 eggs and juveniles/pot. During the fall, 10 and 100 eggs and juveniles were not used hence the threshold for this test could have possibly, been similar to the summer threshold level. Generally, defoliation increased yield loss when combined with nematodes (Table 14). The influence of defoliation level on yield was not as drastic as expected. It is not apparent why defoliation had this slight effect on yield. This is not, however, in agreement with the general principle that nematodes predispose plants to diseases and other pests. Statistical analysis showed that defoliation and M. incognita acted independently in influencing yield.

The gross dollar values had a trend similar to that of yield since they were computed from gross yield. These values are gross figures from which one has to deduct production costs which include pesticides, land rent, labor, interest on loans (if any), and farm machinery depreciation. Thus, net income would depend on the cost of production and current market prices of snap beans. Snap bean production is costly (Taylor and Wilkowske, 1984). Loss in gross income ranges compared to nematode free plants were $470 to $1443; $441 to $2441; $521 to $2059; $764 to $2167; $151 to $2201; and $286 to $2192 when plants were defoliated at various levels at the primary leaf, first trifoliate leaf,

third trifoliate leaf, flower bud formation, full bloom, and pod set stages respectively. If plants were not defoliated but were inoculated with nematodes, loss in income ranged from $443 to $1577; $323 to $1911; $464 to $1827; $855 to $2137; $529 to $1561; and $674 to $1565 when plants were meant to be defoliated at the primary leaf, first trifoliate leaf, third trifoliate leaf, flower bud formation, full bloom, and pod set stages respectively. In each case the lower loss in income is for the 1,000 eggs and juveniles/pot population level and the higher value in loss was for the 100,000 eggs and juveniles/pot (Table 15). These losses are based on yield loss disregarding production costs. The loss in income has been computed using the high market price of snap beans. The yields of snap beans in both studies were low, and thus, the grower would have had a loss in income in both cases.

CHAPTER V
THE EFFECT OF BEAN RUST, UROMYCES PHASEOLI (PERS.) WINT., ON SNAP BEANS, PHASEOLUS VULGARIS L. 'Sprite'

Introduction

Bean rust, caused by the fungus Uromyces phaseoli (Pers.) Wint., is a serious disease of beans, Phaseolus vulgaris L. (Agudelo, 1980; Allen, 1983, McMillan et al., 1982, Pohronezny et al., 1984). The disease causes severe damage on winter and spring grown snap beans in south Florida (McMillan, 1982; Pohronezny et al., 1984). Usually, U. phaseoli first appears in January and becomes progressively more severe February through May (Pohronezy et al., 1984). Initial inoculum is believed to come from infected bean plant debris in abandoned fields. Losses of up to 78% in pinto beans, 74.2% and 18.4% in 'Ex Rico 23' and 'Bat 308' field beans, respectively, have been reported from severely infected crops in the United States and Latin America (CIAT, 1983; Kelly, 1982).

Bean rust, U. phaseoli, is an autoecious polycyclic disease whose rates of increase are affected by timing, amount of sporulation, light intensity, relative humidity, and relative cultivar susceptibility (Cohen and Rotem, 1970; Cook, 1978; Imhoff et al., 1982 a,b). Rotem et al. (1973) reported that, in an automatic humidity chamber study, humidity was inversely related to the sporulation of U. phaseoli. Infection by U. phaseoli has, however, been reported to be favored by prolonged periods of at least 95% relative humidity and moderate temperatures (15-27°C) (Augustin et al., 1972; Gonzalez, 1976; Schein, 1961).

Uromyces phaseoli progress on artificially inoculated beans, P. vulgaris 'Bountiful', depended more on length and frequency of wetting periods than on temperature (Imhoff et al., 1982a).

Yield loss due to disease has been observed to be proportional to the area under the disease progress curve or proportional to disease severity at some critical stage of host growth (Madden et al., 1981; Raymundo and Hooker, 1981; Romig and Calpouzos, 1970; Shaner and Finney, 1977; Teng et al. 1979). In many of these studies, area under the disease progress curve satisfactorily explained the relationship between diseases and yield losses. Disease severity at one or more points in time and rate of increase of the disease were also satisfactory disease parameters employed to explain the relationship between disease and yield loss (James, 1974; James and Teng, 1979; Main, 1977).

Berger (1981) compared the logistic and Gompertz models for disease progress curve fitting. It was observed that the Gompertz model consistently gave better fit to the data examined than the logistic model for disease severity values outside the $0.05 < y < 0.6$ range (Figure 14). The Gompertz model was superior to the logistic model in linearizing 113 selected disease progress curves (Berger, 1981).

Growers often resort to routine fungicide sprays for disease control. Currently, weekly sprays with mancozeb are applied for disease control on beans (McMillan et al., 1982; Pohronezny et al. 1984). The effectiveness of these sprays depends on spray coverage and disease severity but in many cases disease control is less than satisfactory (McMillan et al. 1982). Usually, sprays are initiated before disease signs and/or symptoms are observed on the crop.

The present studies were conducted to determine the effect of bean rust on 'Sprite' snap beans under field conditions.

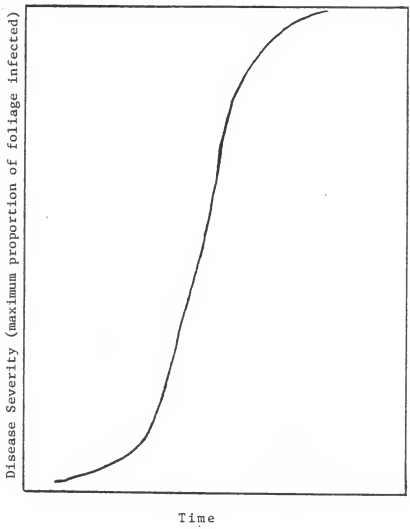


Figure 14. General progress curve of a plant disease.

Materials and Methods

Two trials were conducted at the Tropical Research and Education Center in Homestead, Dade County, Florida, on Rockdale soil (pH ca.7.8). The first trial was planted on 27 February 1985 and the second on 21 March 1985. Beans were harvested on 25 April 1985 and 13 May 1985 respectively.

In both trials plots were 3 rows wide (0.91 m row spacing) and 3 m long. Beans were planted 7-10 cm apart within the row. Prior to planting the herbicides Treflan(R) (841 g ai/ha) and Dual(R) (1.7 kg ai/ha) were applied to the site. Fertilizer (8:16:16) was applied at 448 kg/ha before planting. Plants were top dressed at 224 kg/ha just before flower bud formation. The crops were sprayed with Ambush(R) (40 g ai/ha) fortnightly for cowpea curculio (Chalcodermus aeneus Boh.) control. Slugs and snails were controlled by Mesurol(R) (200 g ai/ha) pellets. Plants were irrigated using an overhead sprinkler system.

In both trials five treatments were arranged in a randomized complete block design with four replications. Fungicides were used as a tool to manipulate disease levels. Treatments used were (a) no fungicide; (b) bitertanol (57 g ai/ha) at 7-day intervals; (c) mancozeb (0.7 kg ai/ha) tank-mixed with sulfur (4.5 kg ai/ha) at 4-5-day intervals; (d) same as (c) but at 7-day intervals; and (e) same as (c) but at 14-day intervals. All sprays were applied with Helena(R) sticker or Nu Film-17(R) as a spreader/sticker. Bitertanol plots were virtually disease free.

Plants were inoculated at the primary leaf growth stage using infected pole bean leaves collected from abandoned bean fields. The

infected leaves were clipped on to wire stakes 25 cm above ground level. Two stakes were placed in each plot, one on each end, depending on general wind direction. Disease progress was monitored by taking trifoliate leaves at random from each plot once a week. At each sampling occasion leaves were taken from the same relative level within the canopy. Disease severity (proportion of leaf area infected by the disease) was determined using the mathematical model $y = \frac{a-b}{a}$, where y = disease severity, a = area of leaf before cutting out diseased tissue, and b = area of leaf after cutting out diseased tissue. The mean of the 5 trifoliate leaves was the measure used in the final data. Leaf area was determined by a LiCor$^{(R)}$ portable area meter (Model LI-300, Lombdar Instruments Corp).

Disease progress curves are generally sigmoid in shape (Imhoff et al., 1982a). The generalized progress curve is shown in Figure 14. Progress curves of bean rust in these studies were obtained by determining disease severity at weekly intervals as indicated above. Area under the disease progress curve was computed using the general model: $y = \sum_{i=1}^{n} [(X_{i+ni} + X_i)/2] [t_{i+1} - t_i]$ in which y = area under the disease progress curve, x = disease severity at the i^{th} observation, t_i = time (days) at the i^{th} observation, and n = total number of observations. The computations were facilitated by the use of a computer program provided by Dr. R. D. Berger. The computer program employed the following model: y = (((n (x) + n (x + 1))/2) * t (x)) where y = area under the disease progress curve, x = disease severity, n = number of disease severity values, and t = time (days) at which observation is made. The rate of disease progress was determined by using the Gompertz model which consistently gave better fit to the data (Berger, 1981).

Gross dollar values were obtained by multiplying yield (kg/ha) by current market prices of 13.62 kg of snap beans which were $6 (low), $11 (medium), and $20 (high), respectively. Net returns from investment were derived by subtracting the gross dollar values of unsprayed plants and the cost of mancozeb from the values realized from sprayed plants. No net returns for bitertanol spray were computed because this fungicide was experimental and its price was not available.

Data were analyzed by the analysis of variance and regression analysis using the general linear procedure of SAS (Ray, 1982).

Results

Figures 15 and 16 show disease progress in trials 1 and 2 respectively. Progress patterns were similar in both trials although disease severity values were higher for trial 2.

Analysis of variance of yield data by treatments gave F values of 5.37 and 10.77 with probabilities of 0.01 and 0.005 for trials 1 and 2, respectively. This showed that there was a significant relationship between yield and disease severity. The disease free plants produced higher yield in trial 2 than in trial 1 (Table 16). In trial 1 disease severity ranged from 0.098 to 0.76 and yield was from 1102 g to 2723 g/plot whereas in trial 2 disease severity ranged from 0.4 to 0.86 and yield was 276 g to 3214 g (Table 16). Generally, where the disease occurred, yield was lower in trial 2 than in trial 1.

Figure 17 shows the relationship between yield loss and maximum proportion of foliage infected (disease severity) for both trials. In trial 1, 0.098, 0.46, 0.65, and 0.76 disease severity resulted in 21%,

17%, 35%, and 60% yield loss respectively whereas in trial 2 disease severity of 0.4, 0.47, 0.71, and 0.86 lead to 55%, 56%, 80%, and 91% yield loss respectively. Figure 18 is a representation of the relationship between area under the disease progress curve (AUDC) and yield for trials 1 and 2. Both disease severity and AUDC were positively correlated with yield loss which showed that these disease measures were inversely related to yield. Figures 17 and 18 are similar in shape. Regression equations between disease severity and snap bean yield shown in Tables 17 and 18 are similar in nature. Regression analysis of the data produced the model $y = a + bx$, y = yield (g/plot), and x = disease parameters. Both disease severity and AUDC were significantly correlated with yield at full bloom and pod set in trial 1 (Table 17). When pods were fully developed only disease severity was significantly correlated with yield. In trial 2, disease severity and AUDC were significantly correlated with yield at pod set through the stage when pods were fully formed (Table 18). In trial 1 the coefficient of determination (R^2) at full bloom or later range from 0.46 to 0.93 (Table 17) while in trial 2 the coefficient of determination (R^2) at pod set or later ranged from 0.89 to 0.99 (Table 18). In trial 1 bean rust severity was not significantly correlated with yield at the stage when pods were half developed whereas the in trial 2 the disease was not significantly related to yield at flower bud formation and full bloom (Tables 17 and 18).

The gross dollar values per hectare of snap beans infected by bean rust are shown in Table 19 and Figures 19 and 20. The virtually disease free plants gave the highest dollar values per hectare in both trials. These plants were sprayed with bitertanol an experiment fungicide, which is currently not registered for rust control on beans. Therefore, no

Therefore, no price information on this product is given. In trial 1, plants with a 0.46 disease severity gave a higher dollar value than plants with a 0.098 disease severity. These dollar values corresponded to spray intervals of mancozeb and sulfur of 7-days and 4-5 days (Table 19). Plants with a disease severity of 0.4 and 0.47 gave dollar values of $2367 and $2327 respectively in trial 2. These disease severity values corresponded with 4-5-day and 7-day spray schedules of mancozeb and sulfur (Table 19). In trial 2, gross dollar values were consistently inversely related to both disease parameters (Table 10). The plants with the highest disease parameter produced the lowest gross dollar value.

The relationship between disease severity and net returns from investment per hectare of snap beans is shown in Table 20. No net returns are shown for the virtually disease free plants because an experimental fungicide with no price tag was used on them. In trial 1 there was no improvement on net returns by spraying beans at 4-5 day intervals from the 7-day intervals. Actually, there was a loss in net income by spraying plants more often (Table 20). In trial 1, there were substantial increases in net returns when plants were sprayed at 7-day intervals compared to the 14-day spray schedule. The increases in returns were $1069, $578, and $306 at the high, medium and low prices at the 7-day spray schedule from the 14-day schedule in trial 1. From the 14-day spray schedule to the 4-5-day spray interval there were increases in net returns of $901, $475, and $239 at the high, medium, and low prices respectively in trial 1. By increasing spray frequency from 7-day to 4-5-day intervals there were net losses of $168, $103, and $67 at the high medium, and low prices respectively. Thus, in trial 1 there

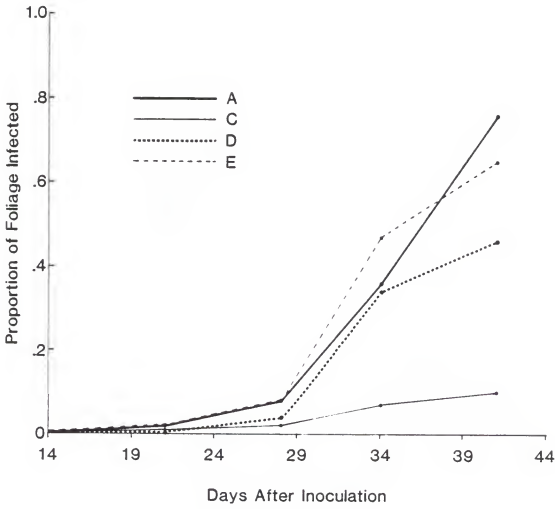


Figure 15. Disease progress curves for Uromyces phaseoli on snap beans sprayed with mancozeb at various frequencies (Trial 1).

A = no fungicide spray, C = mancozeb + sulfur at 4-5 day intervals, D = mancozeb + sulfur at 7-day intervals, and E = mancozeb + sulfur at 14-day intervals.

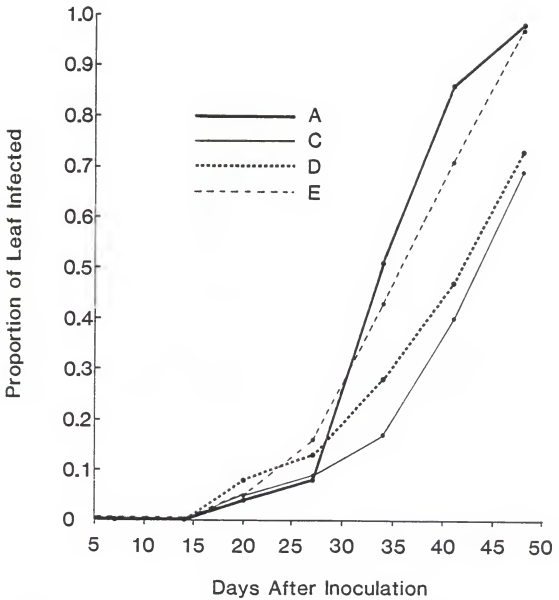


Figure 16. Disease progress curves for Uromyces phaseoli on snap beans sprayed with mancozeb at various frequencies (Trial 1).

A = no fungicide spray, C = mancozeb + sulfur at 4-5 day intervals, D = mancozeb + sulfur at 7-day intervals, and E = mancozeb + sulfur at 14-day intervals.

TABLE 16. Effects of Uromyces phaseoli on snap bean yield.

Disease Parameters					
Maximum Proportion of foliage infested		Area under disease progress curve (sq. units)		Yield (g/plot)	
Trial 1	Trial 2	Trial 1	Trial 2	Trial 1	Trial 2
0.76	0.86	5.64	13.90	1102	276
0.65	0.71	5.93	12.85	1778	642
0.46	0.47	4.11	9.28	2248	1426
0.098	0.40	0.98	7.33	2158	1452
0.0	0.0	0.014	0.0	2723	3214

TABLE 17. Regression equations for the relationship between level bean rust disease and snap bean yield (Trial 1).

Plant growth stage	Regression equation
Third trifoliate leaf	Disease incidence negligible
Flower bud formation	" " "
Full bloom	$y = 2666.71 - 29738.55x$ ($R^2 = 0.9$**)
	$y = 2428.64 - 5481.99d$ ($R^2 = 0.83$*)
Pod set	$y = 2718.09 - 3407.12x$ ($R^2 = 0.93$**)
	$y = 2812.42 - 181.8d$ ($R^2 = 0.84$*)
Pods half developed	$y = 2622.1 - 605.23x$ ($R^2 = 0.69$NS)
	$y = 2441.97 - 1617.92d$ ($R^2 = 0.46$NS)
Pods fully developed	$y = 2618.35 - 196.94x$ ($R^2 = 0.71$NS)
	$y = 2627.26 - 1689.48d$ ($R^2 = 0.8$*)

* R significant at $P \leq 0.05$

** R significant at $P \leq 0.01$

y = yield (g/plot)

x = area under the disease progress curve

d = proportion of foliage infected

NS = not significant at 0.05

TABLE 18. Regression equations for the relationship between bean rust and snap bean yield (Trial 2).

Plant growth stage	Regression equation
First and Third trifoliate	Disease incidence negligible
Flower bud formation	$y = 2492.5 - 7790.18x$ ($R^2 = 0.39$NS)
	$y = 2443.73 - 23678.58d$ ($R^2 = 0.36$NS)
Full bloom	$y = 1940.83 - 1141.86x$ ($R^2 = 0.16$NS)
	$y = 1734.26 - 1384.95x$ ($R^2 = 0.21$NS)
Pod set	$y = 3113.46 - 893.33x$ ($R^2 = 0.89$*)
	$y = 2886.63 - 5340.87d$ ($R^2 = 0.92$**)
Pods half formed	$y = 3099.74 - 369.12x$ ($R^2 = 0.98$**)
	$y = 3220.42 - 3224.52d$ ($R^2 = 0.99$**)
Pods fully formed	$y = 3165.01 - 203.23x$ ($R^2 = 0.98$**)
	$y = 3288.9 - 2795.75d$ ($R^2 = 0.98$**)

* R Significant at 0.05

** R Significant at 0.01

y = yield (g/plot)

x = area under the disease progress curve

d = proportion of foliage infected

NS = not significant

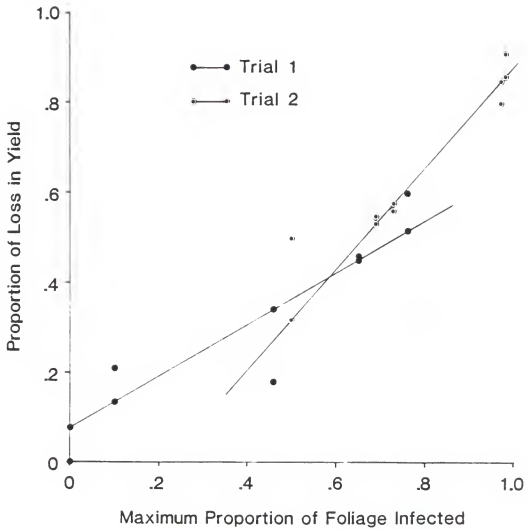


Figure 17. The influence of the maximum proportion of foliage infected by Uromyces phaseoli on snap bean yield.

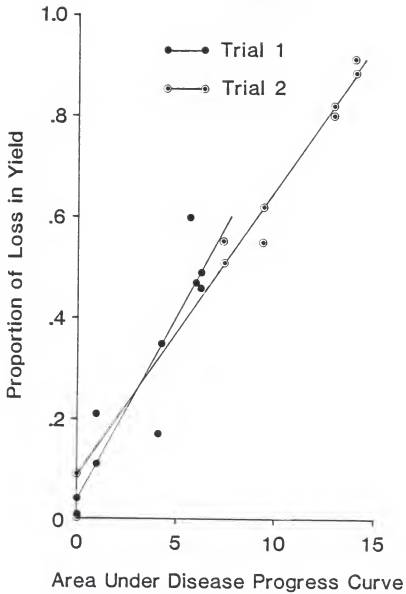


Figure 18. The influence of the area under disease progress curve on snap bean yield.

TABLE 19. The influence of Uromyces phaseoli on the gross dollar value per hectare of snap beans (Phaseolus vulgaris 'Sprite').

Spray frequency (days)	Disease Parameters at harvest: Maximum Proportion of foliage infected		Disease Parameters at harvest: Area under disease progress curve		Price range	Gross dollar values	
	Trial 1	Trial 2	Trial 1	Trial 2		Trial 1	Trial 2
0	0.76	0.86	5.64	13.9	high	1797	449
					medium	987	247
					low	539	135
14	0.65	0.71	5.93	12.85	high	2574	1047
					medium	1416	576
					low	772	314
7	0.46	0.47	4.11	9.28	high	3665	2327
					medium	2016	1280
					low	1100	698
4-5	0.098	0.4	0.98	7.33	high	3520	2367
					medium	1936	1302
					low	1056	710
7[a]	0.0	0.0	0.014	0.0	high	4446	5244
					medium	2446	2884
					low	1334	1573

[a] Sprayed with bitertanol.

TABLE 20. The relationship between disease severity and net return per hectare of snap beans sprayed with mancozeb and sulfur.

No. of sprays	Fungicide spray frequency		Maximum Proportion of foliage infected		Price range	Net return ($) in investment	
	Trial 1	Trial 2	Trial 1	Trial 2		Trial 1	Trial 2
0	0	0	0.76	0.86	high	0	0
					medium	0	0
					low	0	0
3	14-day	14	0.65	0.71	high	743	564
					medium	395	295
					low	199	145
5(6)[a]	7-day	7	0.46	0.47	high	1812	1810
					medium	973	965
					low	505	495
7(8)	4-5-day	4-5	0.098	0.4	high	1644	1828
					medium	870	965
					low	438	485

[a] Number of sprays in trial 2 are in parentheses.

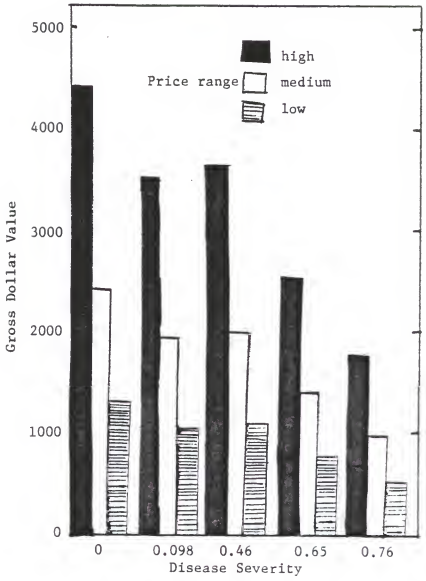


Figure 19. Influence of disease severity on gross dollar value per hectare of 'Sprite' snap beans in trial 1.

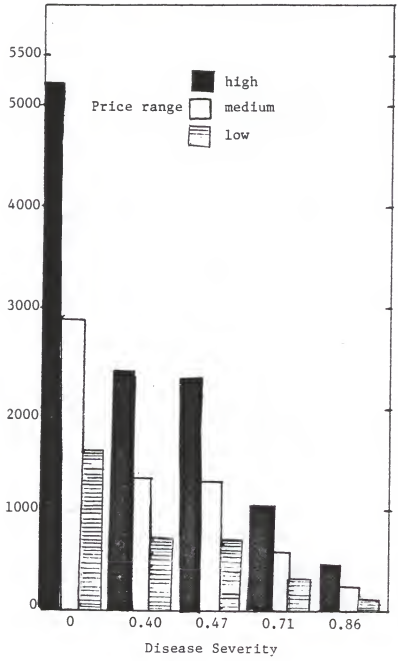


Figure 20. Influence of disease severity on gross dollar value per hectare of 'Sprite' snap beans in trial 2.

was no benefit from spraying the plants more often than 7-day intervals. By spraying at 4-5-day intervals there was a net loss in returns compared to the 7-day intervals. The total cost per spray occasion per hectare was $11.29.

In trial 2, there were increases in net returns, by spraying plants at 7-day intervals compared to the 14-day spray schedule, of $1246, $670, and $350 at the high, medium, and low prices. From the 14-day to the 4-5 day spray schedules, there were increases in net returns of $1264, $670, and $340 at the high, medium, and low prices. When plants were sprayed at 7-day and 4-5-day intervals there was an increase in net returns of $22 and $10 at the high and low prices but there was no net increase in returns at the medium price. Thus, in trial 2 there was no apparent benefit in spraying plants at 4-5-day intervals. The additional sprays hardly paid for the extra cost of spraying more often.

Discussion

Significant relationships were found between yield and both disease severity and area under the disease progress curves. Yields were generally higher in trial 1 than in trial 2. The disease was clearly established earlier in trial 2 than in trial 1.

The relationship between yield or loss in yield and bean rust was well described using both parameters: maximum severity and area under the disease progress curve (AUDC). Rate of disease progress was as well correlated with yield as disease severity or AUDC in both trials with R^2 of 0.71 and 0.79 for trial 1 and trial 2 respectively. In trial 2, disease severity and AUDC had coefficients of determination (R^2) of 0.98

at the same growth stage. This discrepancy in correlation of rate of disease progress and yield in these trials may be due to the early onset of the disease in trial 2 where the rate of disease progress may have started to decrease since most of the foliage had already been infected. There is also a possibility that rates of disease progress were sporadic in trial 2. Although AUDC was an equally good predictor of yield at full bloom and pod set in trial 1 and at pod set through fully formed pods in trial 2, it was a difficult parameter to use since it required the use of computers. Computers are not readily accessible to extension specialists who work closely with local farmers. Disease severity, on the other hand, can be estimated by the use of graph papers or tracing paper for the determination of leaf area before and after cutting out diseased leaf tissue. Thus, disease severity would be a more convenient parameter for the description of the relationship between bean rust and snap bean yield. Moreover, AUDC is derived from disease severity. Area under the disease progress curve is, however, a better representation of the magnitude of the disease for the growing season. Disease severity is, however, easier to measure and does not require complicated calculations. Therefore, disease severity may be useful for the prediction of yield at specific bean plant growth stages.

The Horsfall-Barratt disease rating system was not used in these trials because of its subjectivity. This system has a lot of variability according to the person assessing the disease and would require the comparison of several people's data to be relied on. The Horsfall-Barratt method could, however, be adapted based on standardized diagrams put out by extension.

At disease severity levels (maximum proportion of foliage infected) below 0.5 (50%), it was not worth the expense of the shorter 4-5-day

mancozeb + sulfur spray interval in both trials. The grower would end up losing income by spraying at this frequency. There was no apparent explanation for the slight decrease in yield at the 0.098 disease severity level in trial 1 (Table 16). The decrease in yield may have been caused by adverse effects of the fungicide mixture on plants. The fungicides used to manipulate the disease had zinc and sulfur which are required by plants for proper growth. It was, however, not clear whether the adverse effect was due to over supply of these elements or due to the interaction of these elements and factors. This phenomenon may have been a chance effect which requires further investigation under similar growing conditions.

In trial 2, yield was consistently inversely related to disease severity and AUDC. In this trial lower disease severities were correlated better with higher yields than the higher disease severities. The lower disease severity corresponded with more frequent sprays of mancozeb + sulfur. These low disease parameters resulted from more frequent sprays which cost more money. Thus, returns were reduced substantially. Generally, the shorter 4-5-day mancozeb + sulfur intervals were more costly than the 7-day intervals and the extra cost did not produce enough yield to pay for itself. The optimum spray schedule for beans was, therefore, every 7 days.

From these trials, the grower would be ill-advised to spray snap beans at 4-5-day intervals if the disease severity is below 0.1 and the onset of the disease is late. The economic threshold of bean rust severity was below 0.1 since 0.098 disease severity resulted in 21% yield loss.

There are many methods of determining disease severity in the field. These methods include the use of leaf area meters, the Horsfall-Barratt disease rating system, and pictorial keys. In these trials it has been shown that the use of leaf area is a more convenient method for disease severity assessment. Hence, in order to determine disease severity the grower or extension specialist would have to take random leaf samples at specific intervals and estimate the proportion of foliage infected at each stage. Leaf area determination would be facilitated if a leaf area meter were available but where this was not the case then graph or trace paper could be used. When computers are accessible to the grower/extension specialist then AUDC can be determined based on disease severity. The choice of the variable would depend on availability of expertise and equipment.

Yields were inversely related to disease parameters at specific bean plant growth stages. Rate of disease progress, AUDC and disease severity correlated well with yield. Therefore, the extension worker or grower can choose the parameter of disease to measure. These disease parameters are, however, time consuming which may increase labor and other costs.

CHAPTER VI
THE EFFECT OF DEFOLIATION, METAM-SODIUM, AND BEAN RUST ON SNAP BEANS

Introduction

Beans, Phaseolus vulgaris L., are subject to defoliation by a wide range of factors including insects, diseases, adverse environmental conditions, mammals, and farm machinery (Agudelo, 1980; Allen, 1983; Cook, 1978; Costa and Rossetto, 1972; Ruppel and Idrobo, 1962; Vargas, 1980). The extent to which these factors influence yield depends on the plant growth stage at which they are attacked. Among the most important insect pests feeding on bean leaves are leafminers (Liriomyza spp.), cabbage loopers ((Hub.) Trichoplusia ni), leafrollers (Urbanus proteus L.), Mexican bean beetles (Epilachna varivestis Muls.), and Chrysomelid beetles (Schoonhoven and Cardona, 1980). Bean rust, Uromyces phaseoli (Pers.) Wint., is one of the most damaging diseases of bean in bean producing regions of the world (Acland, 1971; Allen, 1983; Cook, 1978; Crispin and Dongo, 1962; Iraneta and Rodrigez, 1983; Martinez, 1983; Schwartz et al., 1979; Stoetzer and Omunyin, 1983; Vargas, 1980). Other diseases which affect beans include anthracnose (Colletrotrichum lindemuthianum (Sacc. and Mgr.) Bri. and Cav.), angular leaf spot (Isariopsis griseola Sacc.), halo blight, (Pseudomonas syringae pu. phaseolicola (Burkh.) Young, Dye and Wilkie, common blight (Xanthomonas campestris pu. phaseoli (Smith) Dye, and bean common mosaic virus (Acland, 1971; Allen, 1983; Martinez, 1983; Stoetzer and Omunyin, 1983). Root rots caused by Rhizoctonia solani Kuhn, Macrophomina phaseolina

(Tassi) Gold, Sclerotium rolfsii Sacc., Pythium spp., and Fusarium spp. have also been reported to initiate wilting and eventually defoliation (Martinez, 1983). Many nematode species are found in association with bean roots in various parts of the world (Agudelo, 1980, Allen, 1983). The root-knot nematode complex (Meloidogyne spp.) is among the most damaging on beans (Agudelo, 1980; Allen, 1983; Ngundo, 1977; Ngundo and Taylor, 1974, 1975a,b). Thus, an understanding of the relationship between these factors and yield is a prerequisite for the development of a sound pest management strategy.

Information on the relationship between leaf damaging pests and yield has been obtained through pest damage simulation by manually defoliating plants at various growth stages and at several defoliation levels (Edje and Mughogho, 1976a,b; Edje et al., 1972, 1973, 1976; Galvez et al., 1977; Greene and Minnick, 1967; Hohmann and De Carvalho, 1983; Vieira, 1981; Waddill et al., 1984). Ruesink and Kogan (1975) observed that manual defoliation is not precise in simulating pest damage. The imprecision in pest damage simulation may be due to the exact timing of manual defoliation and careful determination of the proportion of the foliage to be removed, which pests cannot do. Moreover, manual defoliation does not introduce saliva and possible phytotoxins which may be important factors in the plant damage caused by specific pests.

Rarely are crop plants attacked by one pest species only. More often several species attack a crop at the same time. Thus, McSorley and Waddill (1982) studied the effect of insect and nematode pests on squash (Cucurbita pepo L.). They partitioned yield loss into insect and nematode components by using multiple regression procedures. It was

observed that the prediction of yield loss was more accurate when both insect and nematode pests were present. Bookbinder and Bloom (1980) reported that Meloidogyne spp. interacted with bean rust, Uromyces phaseoli, on beans. The root-knot nematodes and the disease had an additive effect on the suppression of shoot and root weights of bean plants. Meloidogyne incognita infections reduced uredial diameter of U. phaseoli. Similar effects were observed if U. phaseoli was inoculated first. Bookbinder and Bloom (1980) observed that rusted plants had 62% less M. incognita than uninfected plants. They suggested this was due to suppressed translocation of photosynthates to the roots. U. phaseoli infection did not affect M. incognita egg hatch.

This study was conducted to determine the relationship between defoliation, nematodes and bean rust and yield of snap beans.

Materials and Methods

General

Three trials were conducted at the Tropical Research and Education Center in Homestead, Dade County, Florida. Experimental sites were on Rockdale soil (pH ca. 7.8) planted in fall 1984 and early spring 1985. The fields had been previously cropped in tomato (Lycopersicon esculentum Mill.). In all three trials snap beans (Phaseolus vulgaris L. 'Sprite') were used. Prior to planting, the herbicides Treflan(R) (841 ai/ha) and Dual(R) (1.7 kg ai/ha) were applied to the site. Fertilizer (8:16:16) was applied preplant at 448 kg/ha according to the University of Florida Extension recommendation (Stall and Sherman, 1983). The plants were topdressed with 224 kg/ha fertilizer at flower

bud formation. Metam-sodium was used to manipulate nematode populations. Subsequent weeding was done by cultivation. Irrigation was provided by an overhead sprinkler system.

The effect of metam-sodium on snap beans

The crop was planted on 26 November 1984. Individual plots consisted of 3 rows 3m long with 0.91m between rows. Seeds were planted at 8-10 cm spacing. Treatments were replicated 4 times in a randomized complete block. Metam-sodium was applied preplant at 0, 47, 94, 187, 281, and 374 L/ha. Preplant soil samples consisted of a composite mixture of 10 soil scoops (to a 6-8 cm depth) from each plot 12 days after fumigation. The 12-day period was based on the observations made by McSorley and Parrado (1984). Aliquots of 100 ml soil were processed by sieving and centrifugal flotation (Jenkins, 1964). Subsequent soil samples at mid-season and harvest were taken from the root zone and similarly extracted. Only live nematodes were counted in the preplant samples but in the later samples, nematodes were first killed by gentle heating in a water bath (55-60°C) and counted. Beans were harvested on 31 January 1985.

Yield data were subjected to analysis of variance and regression analysis using the general linear models procedure of SAS (Ray, 1982).

The effect of metam-sodium and defoliation on snap beans

The crop was planted on 26 November 1984. Individual plots consisted of 4 rows 3 m long with 0.91 m between rows. Seeds were planted at 8-10 cm spacing within the row. A split-plot design was used in this trial to investigate the effect of metam-sodium (main plots) and defoliation (subplots) on snap beans. Treatments were replicated 4 times. Each subplot consisted of a 3 m long row. Defoliation levels

investigated were 0%, 25%, 50%, and 75% and metam-sodium was applied at 0, 47, 94, 187, and 374 L/ha. Metam-sodium was applied preplant. Preplant soil samples were taken 12 days after fumigation by compositing 10 soil scoops (to a depth of 6-8 cm) from each main plot. Aliquots of 100 ml soil were processed by sieving and centrifugal flotation. Subsequent soil samples were taken at midseason and harvest from the root zone and similarly treated. Only live nematodes were counted from the preplant soil samples and in the later samples nematodes were killed before counting.

Defoliation was accomplished by removing the lamina from the distal end of the petiole using a pair of scissors. Plants were defoliated once at flower bud formation. Beans were harvested on 31 January to 1 February 1985.

Data were analyzed by analysis variance and covariance followed by regression analysis using the general linear models procedure of SAS (Ray, 1982).

The effect of defoliation, metam-sodium and bean rust on snap beans

A 4x3x2 factorial experiment was conducted at the Tropical Research and Education Center in Homestead, Dade County, Florida, on Rockdale soil (pH ca. 7.8). The crop was planted on 26 March 1985. Plots consisted of 3 rows, 3 m long with 0.91 m between rows. Seeds were planted at 8-10 cm spacing within the row. Fertilizer (8:16:16) was applied preplant at 448 kg/ha and plants were topdressed at flower bud formation at 224 kg/ha.

Metam-sodium was applied at 935 L/ha and a fumigated control was included. Defoliation levels investigated were 0, 25%, and 50%. Bean rust was manipulated by sprays of bitertanol (57 g ai/ha) at 7-day

intervals, mancozeb (1.7 kg ai/ha) plus sulfur (4.5 kg ai/ha) at 7-day and 14-day intervals. A no-spray plot was included to ensure a high disease level at specific times of assessment. Helena(R) sticker or Nu Film(R)-17 was used as a surfactant in all fungicide sprays.

Preplant soil samples were taken by compositing 10 soil scoops from each plot 12 days after fumigation. Aliquots of 100 ml soil were processed by sieving, and nematodes were extracted by centrifugal flotation (Jenkins, 1964). Subsequent soil samples taken at mid season and at harvest were similarly handled. Only live nematodes were counted in the preplant soil samples. In the later samples the nematodes were first killed by heating in a water bath and then counted.

Plants were subjected to rust inoculum at the primary leaf stage by clipping infected pole bean leaves on to wire stakes just east of the test plots and 25 cm off the ground. Disease progress was monitored by taking 5 trifoliate leaves from each plot once a week. The leaf area before and after cutting diseased leaf tissue was determined. The leaf samples were taken from the same general position in the canopy at each sampling.

For the defoliation treatments, plants were manually defoliated with pairs of scissors. The foliage was removed from the distal end of the petiole. Defoliation was done only once, at full bloom because results from other workers indicated that this was a critical growth stage for beans (Hohmann and DeCarvalho, 1983).

Data were subjected to analysis of variance and regression analysis using the general linear models procedure of SAS (Ray, 1982).

Results

Effect of metam-sodium on snap beans

Tables 21, 22 and 23 show the numbers of nematodes in 100 ml aliquots of soil at preplant, mid-season and harvest respectively. Two nematode genera, Criconemella and Rotylenchulus, were found in the soil at preplant (Table 21). At mid-season, four nematode genera were detected in the soil: Helicotylenchus, Meloidogyne, Rotylenchulus, and Tylenchorhynchus (Table 22). Meloidogyne had the highest numbers in unfumigated plots (Table 22), but at this time, the effect of metam-sodium on nematode population was not proportional to its rate of application. Criconemella, Helicotylenchus, Meloidogyne, and Rotylenchulus were found in the soil at harvest (Table 23). Metam-sodium had no significant effects on nematode numbers at any sampling (Tables 21, 22, and 23).

Analysis of variance on snap bean yield showed that there were significant differences among metam-sodium rates ($F = 3.4^*$). Yield responses were, however, not consistently proportional to metam-sodium rates (Table 21). Regression analysis on the relationship between metam-sodium and yield produced models of the form $Y = -0.004x^2 + 345.75$ ($R^2 = 0.17$) and $Y = -0.00004x^3 - 0.03x^2 + 4.9x + 287.5$ ($R^2 = 0.22$) where Y = yield (g/plot), and x = metam-sodium rate (L/ha). The cubic model gave a higher coefficient of determination than the quadratic model (Figure 21). The coefficients of determination were very low. The linear model of the form $Y = a + bX$ gave an R^2 of only 0.12 indicating that yield was not linearly related to metam-sodium rate. Multiple regression on nematode general effects on yield gave the model $Y = a + b_1 x_1 + b_2 x_2 + \ldots + bn\ xn$ where Y = yield, x = log (nematode population

+ 1). The linear models gave low R^2 at all soil sampling times (Table 24).

Gross dollar values per hectare are shown in Table 25. The highest dollar value was obtained with the 374 L/ha soil fumigation, as expected, since this was the rate at which the highest yield was achieved. The gross dollar values were based on yield per hectare and the following prices \$6.00 (low), \$11.00 (medium), and \$20.00 (high) per bushel of snap beans (13.62 kg). Net income is shown in Table 26. Yields obtained in this study were generally low. These yields were low probably because of the less ideal temperatures at the time the study was conducted. Under these conditions the grower would have made a profit at 47 L/ha, and 94 L/ha at the low, medium, and high snap bean prices respectively (Table 26). The dollar values were based on the metam-sodium cost of \$1.59/L (McSorley and Pohronezny, 1984); no other expenses have been used in economic analyses.

The effect of metam-sodium and defoliation on snap beans

Analysis of variance on the influence of metam-sodium, defoliation, and their interaction on snap beans showed that there was no significant interaction between metam-sodium and manual defoliation (Table 27). There were also no significant differences among metam-sodium rates (F = 1.5 NS) on yield. There were, however, significant differences among defoliation levels (F = 20.22**) (Table 27). Regression analysis of yield data on metam-sodium rates produced the model $y = a + bx - cx^2$ (R^2 = 0.34) where y = yield (g/plot), x = metam-sodium rate (gal/acre) (Figure 21). The cubic model of the form $y = a + bx + cx^2 + dx^3$ (R^2 = 0.46) was obtained when yield data were analyzed by regression against manual defoliation (Figure 28).

TABLE 21. Effect of metam-sodium on snap bean yield and populations of nematode genera in preplant soil samples. Data are means of 3 replicates.

Metam-sodium (Liters/ha)	*Criconemella* (No./100 ml)	*Rotylenchulus* (No./100 ml)	Yield (g/plot)	Yield loss (%)
0	1	21	360	49
47	1	27	596	16
94	4	72	650	8
187	1	17	680	4
281	0	42	581	18
374	0	25	707	0

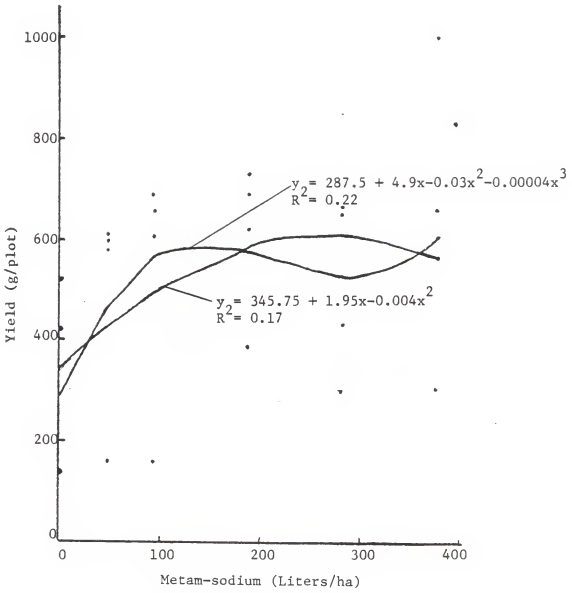


Figure 21. Effect of metam-sodium on snap bean yield.

TABLE 22. Effect of metam-sodium on snap bean yield and populations of nematode genera in midseason soil samples. Data are means of 3 replicates.

Metam-sodium (Liters/ha)	Criconemella (No./100 ml)	Helicotylenchus (No./100 ml)	Meloidogyne (No./100 ml)	Rotylenchulus (No./100 ml)	Tylenchorhynchus (No./100 ml)	Yield (g/plot)
0	0	3	12	0	4	360
47	0	8	8	0	8	596
94	2	4	0	1	0	650
187	0	1	7	0	1	680
281	0	3	11	0	5	581
374	0	1.3	3	1	2	707

TABLE 23. Effect of metam-sodium on snap bean yield and populations of nematode genera in final soil samples (at harvest).

Metam-sodium (Liters/ha)	Criconemella (No./100 ml)	Helicotylenchus (No./100 ml)	Meloidogyne (No./100 ml)	Rotylenchulus (No./100 ml)	Yield (g/plot)
0	1	21	4	25	360
47	1	27	6	1	596
94	4	72	1	4	650
187	1	17	0	0	680
281	0	42	0	18	581
374	0	25	0	1	707

TABLE 24. Regression equations for the relationship between various nematode populations, during the growing season, and snap bean yield. Nematode populations were transformed to log (population +1).

Time	Regression Equation[a]	Coefficient of determination (R^2)
Preplant	$y = 497.3-21x_1+7.1x_4$	0.025 NS
	$y = 538.4-20.3x_1$	0.018 NS
Midseason	$y = 894.6-191.1x_1-11.1x_2-25.4x_3-66.6x_4-8.2x_5$	0.17 NS
	$y = 902.7-200.6x_1-12.3x_2-29.6x_3-67.2x_4$	0.17 NS
	$y = 841.7-220.7x_1-24.8x_2-20.3x_3$	0.14 NS
	$y = 767.7-187.2x_1-30.4x_2$	0.13 NS
	$y = 770.4-251.61x_1$	0.11 NS
Harvest	$y = 797.3-157.6x_1-35.5x_2-28.6x_3+9.4x_4$	0.17 NS
	$y = 783.5-142.8x_1-27.1x_2-25.8x_3$	0.17 NS
	$y = 731.7-132x_1-27.3x_2$	0.15 NS
	$y = 682.8-137x_1$	0.13 NS

[a] X_1 = Criconemella

X_2 = Helicotylenchus

X_3 = Meloidogyne

X_4 = Rotylenchulus

X_5 = Tylenchorhynchus

NS = Not significant at 0.05

TABLE 25. The effect of metam-sodium on gross dollar values per hectare of beans.

Vapam (liters/ha)	Price Range		
	low	medium	high
0	176	323	587
47	292	535	972
94	318	583	1061
187	333	610	1094
281	285	522	948
374	346	634	1153

TABLE 26. Net returns on investment per hectare of snap beans.

Metam-sodium (Liters/ha)	Price Range		
	low	medium	high
0	0	0	0
47	41	137	311
94	- 7	112	325
187	-141	- 10	210
281	-338	-247	- 85
374	-425	-283	- 29

TABLE 27. F-values from analysis of variance for the effects of metam-sodium, defoliation and their interaction on snap bean yield.

Source	F	Probability of F
Metam-sodium	1.5	0.22
Defoliation	20.22	0.0001
Defoliation x metam-sodium	0.62	0.82

TABLE 28. Nematode genera found in soil samples collected (preplant). Data are means of 4 replicates

Metam-sodium (Liters/ha)	Criconemella (No./100 ml)	Helicotylenchus (No./100 ml)	Rotylenchulus (No./100 ml)	Tylenchorhynchus (No./100 ml)
0	0	0	4	4
47	0	1	6	2
94	0	3	0	2
187	0	0	2	.0
374	1	0	2	0

TABLE 29. Nematode genera found in soil samples (midseason). Data are means of 4 replicates.

Metam-sodium (Liters/ha)	Helicotylenchus (No./100 ml)	Meloidogyne (No./100 ml)	Rotylenchulus (No./100 ml)
0	3	15	5
47	3	19	3
94	4	15	0
187	8	19	4
374	0	19	3

TABLE 30. Nematode genera found in soil samples (harvest). Data are means of 4 replicates.

Metam-sodium (Liters/ha)	Criconemella (No./100 ml)	Helicotylenchus (No./100 ml)	Meloidogyne (No./100 ml)	Rotylenchulus (No./100 ml)	Tylenchorhynchus (No./100 ml)
0	0	8	4	9	0
47	0	5	8	9	1
94	2	2	6	9	1
187	0	2	5	9	1
374	0	0	3	4	0

TABLE 31. Mean snap bean yield of plants defoliated at various levels on plots fumigated with metam-sodium.

Defoliation level (proportion of foliage)	Metam-sodium (Liters/ha)	Yield (g/plot)	Yield loss (%)[a]
0	0	545	36
0	47	551	35
0	94	738	13
0	187	851	0
0	374	568	33
0.25	0	469	45
0.25	47	417	51
0.25	94	596	30
0.25	187	403	53
0.25	374	423	50
0.50	0	264	69
0.50	47	253	70
0.50	94	371	56
0.50	187	330	61
0.50	374	335	61
0.75	0	263	69
0.75	47	186	78
0.75	94	276	68
0.75	187	194	77
0.75	374	284	67

[a] Yield loss compared to maximum yield of 851 g.

TABLE 32. The effect of defoliation and metam-sodium on the gross dollar value per hectare of snap beans.

Defoliation level (proportion of foliage)	Metam-sodium (Liters/ha)	Gross dollar values by price range		
		Low	Medium	High
0	0	267	489	889
0	47	269	494	898
0	94	361	662	1204
0	187	416	763	1388
0	374	278	510	927
0.25	0	230	421	765
0.25	47	204	374	681
0.25	94	292	535	972
0.25	187	197	362	658
0.25	374	207	279	690
0.5	0	129	237	431
0.5	47	124	227	412
0.5	94	182	333	605
0.5	187	162	296	539
0.5	374	164	301	547
0.75	0	129	236	429
0.75	47	91	167	303
0.75	94	135	247	450
0.75	187	95	174	315
0.75	374	164	255	463

TABLE 33. Net returns on investment ($) per hectare of snap beans. Plants were defoliated at various levels and soil treated with metam-sodium.

Defoliation level (proportion of foliage)	Metam-sodium (Liters/ha)	Net returns by price range		
		Low	Medium	High
0	47	- 27	- 25	- 21
0	94	34	112	255
0	187	29	154	379
0	374	-229	-220	-203
0.25	0	- 37	- 68	-124
0.25	47	- 93	-145	-238
0.25	94	- 35	- 14	- 23
0.25	187	-190	-247	-351
0.25	374	-300	-350	-440
0.50	0	-137	-252	-458
0.50	47	-173	-292	-507
0.50	94	-145	-217	-345
0.50	187	-225	-313	-470
0.50	374	-343	-429	-583
0.75	0	-138	-253	-460
0.75	47	-206	-352	-616
0.75	94	-192	-301	-499
0.75	187	-292	-436	-694
0.75	374	-343	-475	-666

TABLE 34-1. F values from the analysis of variance for yield.

Source	F value	Probability of F
Defoliation	3.06	0.07
Metam-sodium	0.12	0.73
Defoliation x Metam-sodium	2.20	0.12
Fungicide	7.62	0.0002
Defoliation x Fungicide	1.15	0.34
Metam-sodium x Fungicide	1.41	0.25
Defoliation x Fungicide x Metam-sodium	0.62	0.72

TABLE 34-2. Mean snap bean yield per plot sprayed with fungicides.

Fungicide	Spray Frequency	Mean yield (g/plot)[c]
No fungicide	0	284a
Mancozeb	14-day	396a
Mancozeb	7-day	342a
Bitertanol	7-day	616b

[c] Means followed by the same letter are not significantly different at $P \leq 0.05$ (Duncan's multiple range test).

TABLE 35. Regression equations for the effect of defoliation, nematodes, and bean rust on snap beans.

Time of assessment of disease	Regression equation[1]	
Flower bud formation (preplant nematode counts)	$y = 637.7-262.7x_1-6608.9x_2+26.3x_3-62x_4+2.8x_5$	($R^2 = 0.45$**)
	$y = 638.8-332.3x_1-6497.6x_2$	($R^2 = 0.42$**)
	$y = 547.6-6164.1x_2$	($R^2 = 0.26$**)
Full bloom (preplant nematode counts)	$y = 539.6-234.8x_1-3269.5x_2+55.1x_3+25.5x_4-1.32x_5$	($R^2 = 0.67$**)
	$y = 653.56-314.82x_1-299.2$	($R^2 = 0.61$**)
	$y = 572.46-295.5x2$	($R2 = 0.47$**)
Pod formation (preplant nematode counts)	$y = 695.7-290x_1-947.7x_2+7.6x_3-102.6x_4+9.8x_5$	($R^2 = 0.56$**)
	$y = 646.1-348.4x_1-925x_2$	($R^2 = 0.52$**)
	$y = 549.6-870.6x_2$	($R^2 = 0.36$**)
Pods half-developed (preplant nematode counts)	$y = 740.4-275.2x_1-569.1x_2-2.6x_3-78.1x_4+2.1x_5$	($R^2 = 0.62$**)
	$y = 665.9-301.2x_1-564x_2$	($R^2 = 0.61$**)
	$y = 590.4-563.3x_2$	($R^2 = 0.48$**)
Pods fully formed (preplant nematode counts)	$y = 702.8-283.4x_1-398.3x_2+2.2x_3-11.1x_4-2.3x_5$	($R^2 = 0.66$**)

TABLE 35. Continued.

Time of assessment of disease	Regression equation[1]	
	$y = 687.1-288.9x_1-397.7x_2$	($R^2 = 0.66$**)
	$y = 616.7-401x_2$	($R^2 = 0.54$**)
Pods fully formed (final nematode counts)	$y = 702.8-283.4x_1-398.3x_2+2.2x_3-11.1x_4-2.3x_5$	($R^2 = 0.66$**)
	$y = 687.1-288.9x_1-397.7x_2$	($R^2 = 0.66$**)
	$y = 616.7-401x_2$	($R^2 = 0.54$**)
Pods fully formed (final nematode counts)	$y = 869.8-230.4x_1-415.9x_2+6x_3-18.3x_5+6x_6-42.4x_7$	($R^2 = 0.69$**)
	$y = 734.6-282.5x_1-410.7x_2-33x_7$	($R^2 = 0.66$**)
	$y = 616.7-401.4x_2$	($R^2 = 0.54$**)

[a] y = yield (g/plot; x_1 = defoliation level; x_2 = disease severity; x_3 = Heliotylenchus log (x+1); x_4 = Meloidogyne log (x+1); x_5 = Rotylenchulus log (x+1); x_6 = Tylenchorhynchus log (x+1); x_7 = Criconemella log (x+1).

** R significant at $P \leq 0.01$.

TABLE 36. Nematode genera and disease severity values found in plots fumigated with metam-sodium. Figures are means of 3 replicates (Preplant soil samples).

Defoliation level (proportion of foliage)	Metam-sodium (Liters/ha)	Disease severity (maximum proportion of foliage infected)	Helicotylenchus (No./100 ml)	Meloidogyne (no./100 ml)	Rotylenchulus (No./100 ml)
0	0	0.02	0	0	19
0	935	0.05	0	0	16
0.25	0	0.04	5	0	5
0.25	935	0.04	0	0	2
0.50	0	0.03	0	0	3
0.50	935	0.03	5	0	2
0	0	0.01	5	0	36
0	935	0.01	0	0	15
0.25	0	0	5	0	37
0.25	935	0.03	0	0	15
0.50	0	0.0	5	0	37
0.50	935	0.0	0	0	15
0	0	0.02	0	0	13
0	935	0.04	5	0	27
0.25	0	0.02	0	0	9
0.25	935	0.02	0	0	9
0.50	0	0.02	0	5	2
0.50	935	0.5	0	0	12
0	0	0.04	10	0	12
0	935	0.02	10	0	45
0.25	0	0.03	0	0	6
0.25	935	0.02	0	0	3
0.50	0	0.03	10	0	30
0.50	935	0.01	0	0	40

TABLE 37. Nematode genera + disease severity values found in plots fumigated with metam-sodium. Figures are means of 3 replicates (Soil samples at harvest).

Defoliation level (proportion of foliage)	Metam-sodium (Liters/ha)	Disease severity (maximum proportion of foliage infected)	Criconemella (No./100 ml)	Helicotylenchus (No./100 ml)	Rotylenchulus (No./100 ml)	Tylenchorhynchus (No./100 ml)
0	0	0.87	0	13	85	10
0	935	0.83	0	2	77	41
0.25	0	0.64	0	13	93	13
0.25	935	0.71	0	8	32	10
0.5	0	0.77	0	5	134	8
0.5	935	0.74	2	2	47	3
0	0	0.02	2	28	66	23
0	935	0	0	15	77	2
0.25	0	0	2	7	125	15
0.25	935	0.01	2	23	108	25
0.5	0	0	0	2	78	7
0.5	935	0	2	2	83	18
0	0	0.56	2	18	87	7
0	935	0.59	0	13	77	7
0.25	0	0.7	0	5	98	3
0.25	935	0.66	0	17	67	18
0.5	0	0.68	2	5	103	15
0.5	935	0.71	0	8	118	3
0	0	0.71	0	23	135	20
0	935	0.74	0	7	45	5
0.25	0	0.75	2	13	120	3
0.25	935	0.83	2	7	45	12
0.5	0	0.72	0	8	100	17
0.5	935	0.81	0	20	129	28

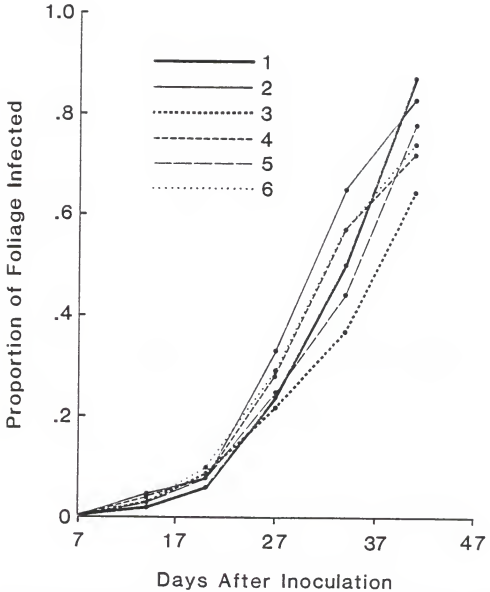


Figure 22. Disease progress curve for Uromyces phaseoli on snap beans not sprayed with mancozeb, defoliated at various levels and soil fumigated with metam-sodium.

1 = no defoliation, no metam-sodium and no fungicide, 2 = no defoliation, 935 L/ha metam-sodium, and no fungicide, 3 = 25% defoliation, no metam-sodium and no fungicide, 4 = 25% defoliation, 935 L/ha metam-sodium and no fungicide, 5 = 50% defoliation, no metam-sodium and no fungicide, 6 = 50% defoliation, no metam-sodium and no fungicide.

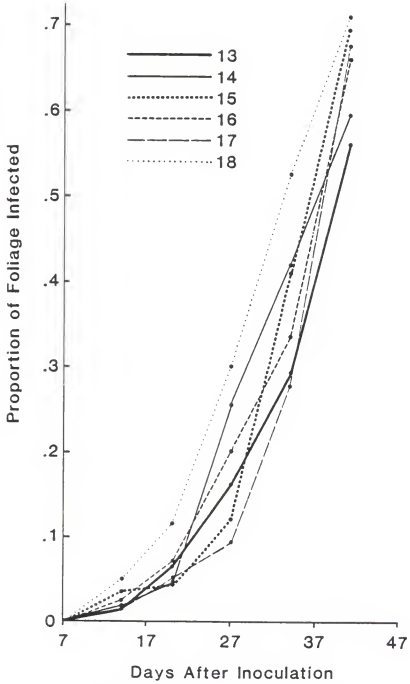


Figure 23. Disease progress curves for Uromyces phaseoli on snap beans sprayed with mancozeb at 7 day intervals, defoliated at various levels and soil fumigated with metam-sodium.

13 = no defoliation, no metam-sodium and mancozeb + sulfur at 7-day intervals, 14 = no defoliation, 935 L/ha metam-sodium and mancozeb + sulfur at 7-day intervals, 15 = 25% defoliation, no metam-sodium and mancozeb + sulfur at 7-day intervals, 16 = 25% defoliation, 935 L/ha metam-sodium and mancozeb + sulfur at 7-day intervals, 17 = 50% defoliation, no metam-sodium and mancozeb + sulfur at 7-day intervals, and 18 = 50% defoliation, 935 L/ha metam sodium and mancozeb + sulfur at 7-day intervals.

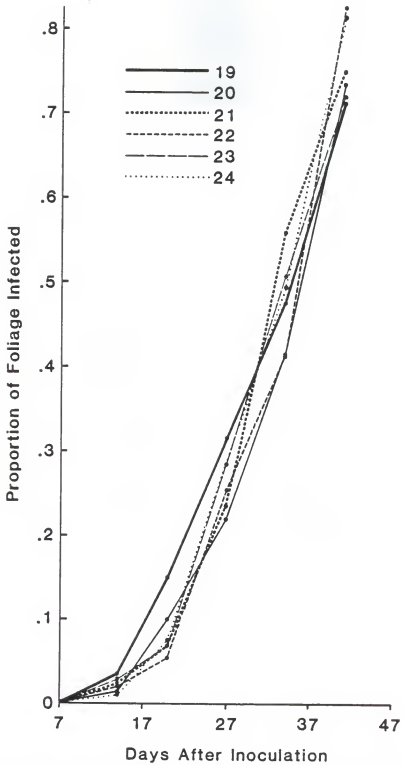


Figure 24. Disease progress curves for Uromyces phaseoli on snap beans sprayed with mancozeb at 14-day intervals, defoliated at various levels and soil fumigated with metam-sodium.

19 = no defoliation, no metam-sodium and mancozeb + sulfur at 14-day intervals, 20 = no defoliation, 935 L/ha metam-sodium and mancozeb + sulfur at 14-day intervals, 21 = 25% defoliation, no metam-sodium and mancozeb + sulfur at 14-day intervals, 22 = 35% defoliation, 935 L/ha metam-sodium and mancozeb + sulfur at 14-day intervals, 23 = 50% defoliation, no metam-sodium and mancozeb + sulfur at 14-day intervals, and 24 = 50% defoliation, 935 L/ha metam-sodium and mancozeb + sulfur at 14-day intervals.

TABLE 38. Effect of defoliation, metam-sodium and bean rust on yield of gross dollar value per hectare of snap beans. Data are means of 3 replicates.

Defoliation level (proportion of foliage)	Metam-sodium (Liters/ha)	Disease severity (proportion of foliage infested)	Yield (g/plot)	Gross dollar values Price Range high	medium	low
0	0	0.87	184	300	165	90
0	935	0.83	365	596	328	179
0.25	0	0.64	260	424	233	127
0.25	935	0.71	345	563	310	169
0.5	0	0.77	220	359	197	108
0.5	935	0.74	338	551	303	165
0	0	0.02	807	1316	724	395
0	935	0	827	1349	742	405
0.25	0	0	570	930	512	279
0.25	935	0.01	499	814	448	244
0.5	0	0	446	727	400	219
0.5	935	0	548	895	492	268
0	0	0.56	349	569	313	171
0	935	0.59	573	935	514	281
0.25	0	0.70	520	848	467	255
0.25	935	0.66	276	450	247	135
0.5	0	0.68	258	420	231	126
0.5	935	0.71	143	233	128	70
0	0	0.71	363	593	326	178
0	935	0.74	406	662	364	199
0.25	0	0.75	190	310	171	93
0.25	935	0.83	359	585	322	176
0.5	0	0.72	327	533	293	160
0.5	935	0.81	397	648	357	196

TABLE 39. Net returns ($) on investment per hectare of snap beans.

Defoliation level (proportion of foliage)	Metam-sodium (Liters/ha)	Disease severity	Net Income Price Range high	medium	low
0	0	0.87	300	165	90
0	935	0.83	-1191	-1324	-1398
0.25	0	0.64	124	68	38
0.25	935	0.71	-1224	-1342	-1408
0.5	0	0.77	59	32	9
0.5	935	0.74	-1236	-1349	-1562
0	0	0.02	1016	589	305
0	935	0	- 438	910	-1172
0.25	0	0	670	346	189
0.25	935	0.01	- 973	-1204	-1332
0.5	0	0	427	235	128
0.5	935	0	- 892	-1160	-1308
0	0	0.56	213	92	81
0	935	0.59	- 908	-1194	-1352
0.25	0	0.7	420	245	108
0.25	935	0.66	-1394	-1461	-1498
0.5	0	0.68	64	10	- 20
0.5	935	0.71	-1610	-1580	-1563
0	0	0.71	263	161	54
0	935	0.74	-1158	-1321	-1412
0.25	0	0.75	- 24	- 28	- 31
0.25	935	0.83	-1235	-1364	-1435
0.5	0	0.72	199	94	36
0.5	935	0.81	-1194	-1451	-1436

Tables 28, 29, and 30 show nematode genera detected in the soil at preplant, midseason, and at harvest. Metam-sodium rate had no significant effects on nematode numbers at all sampling times.

Total yield (g/plot) and yield loss are shown in Table 31. The lowest yield was obtained from plots fumigated at 47 L/ha metam-sodium and plants defoliated at the 0.75 level. Gross dollar values are given in Table 32 and net income is shown in Table 33. These gross dollar values were computed from total yield (per hectare) multiplied by the following price ranges: $6.00 (low), $11.00 (medium), and $20.00 (high) per bushel (13.62 kg). Net dollar values were obtained by subtracting the cost of metam-sodium per hectare from the gross dollar values. Gross dollar values generally decreased with the increase in defoliation level. There were net returns on investment when plants were not defoliated (Table 33). There were no net returns on investment when plants were defoliated at any level (Table 33).

The effect of defoliation, metam-sodium, bean rust, and their interaction on snap bean yield

Analysis of variance showed that there were no significant differences among defoliation levels and metam-sodium rates at the 0.05 level (Table 34-1). There was no significant interaction among defoliation levels, metam-sodium rates, and fungicides at the 0.05 level (Table 34-1). There were, however, significant differences among fungicide sprays ($F = 7.62^{**}$). When analysis of variance was used for yield data analysis with disease severity as one of the independent factors, there were no significant differences in yield based on disease severity (maximum proportion of foliage infected) ($F < 1$). There was no significant interaction among defoliation levels, metam-sodium rates,

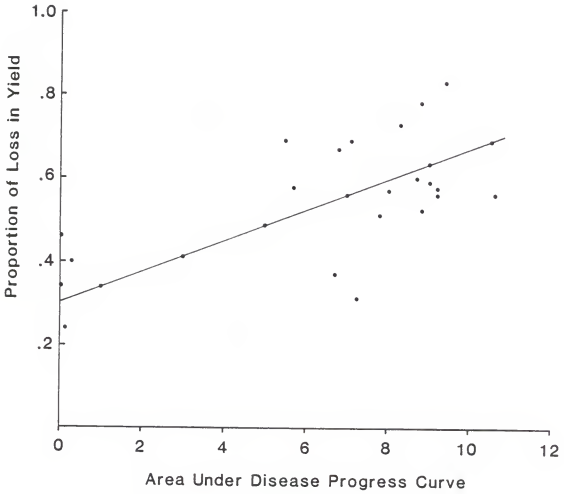


Figure 25. The relationship between area under disease progress curve and yield loss of snap beans.

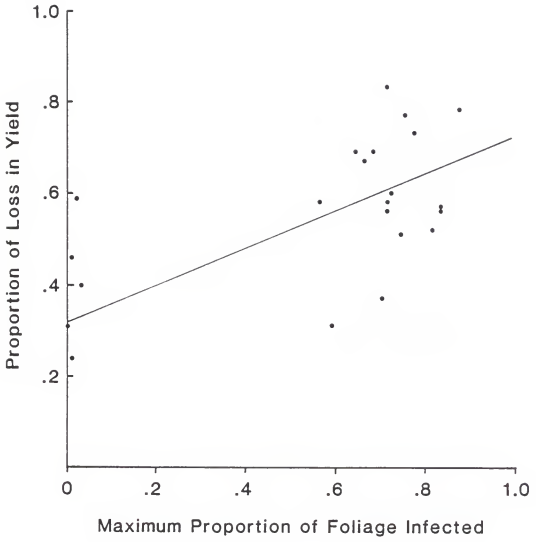
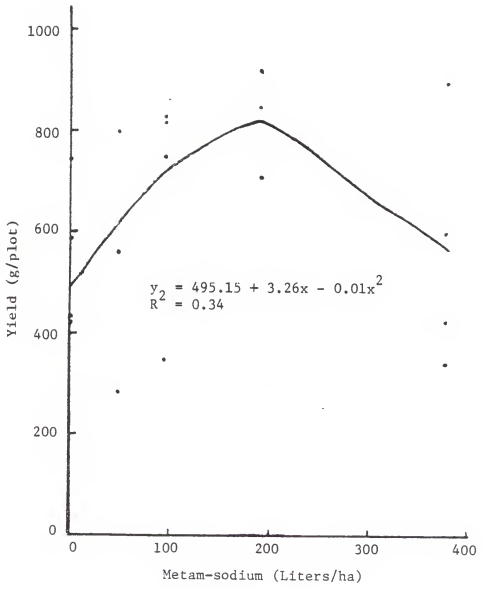


Figure 26. The relationship between yield loss and maximum proportion of foliage infected.



igure 27. The effect of metam-sodium on nsap bean yield when plants were not defoliated.

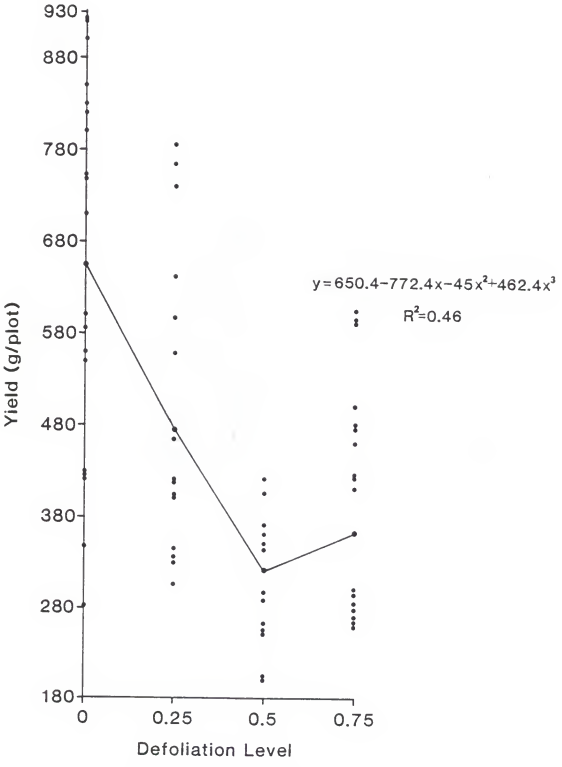


Figure 28. The general relationship between defoliation and snap bean yield at all metam-sodium rates.

and disease severity. Since there were significant differences among fungicides means were separated using Duncan's multiple range test showed that there were no significant differences between mancozeb sprays and the unsprayed treatment. There was, however, significant difference between bitertanol and mancozeb sprays (Table 34-2).

Regression analysis on the effect of defoliation, nematodes, and disease severity on snap bean yield produced models of the form: $y = a + b_1 X_1 + b_2 X_2 + \ldots + bnxn$, where y = yield (g/plot), $x_1 \ldots x_n$ = independent variable (defoliation level, disease severity, and log (nematode population + 1) (Table 35). Stepwise regression showed that disease severity contributed most to the coefficients of determination (R^2) (Table 35). Since there were no significant differences among defoliation levels and metam-sodium rates, further explanations in this section will be restricted to disease severity or area under the disease progress curve (AUDC). Thus, figures 25 and 26 show the relationship between yield loss and AUDC and disease severity, respectively. The relationship between these disease parameters and yield should be the inverse of their relationship with yield loss. Regression analysis of yield data produced models of the form $y = a-bx$, where y = yield (g/plot), x = either disease severity or AUDC (Table 35). The coefficients of determination (R^2) for disease severity and AUDC were 0.54 and 0.52 respectively when the disease was assessed at harvest. Coefficients of determination for disease severity for the other times of disease assessment are given in Table 35. Since disease assessment severity consistently had better R^2 than AUDC, it was generally used in yield data analysis. Moreover, a set of disease severity points results in AUDC. Disease progress curves are shown in figures 22, 23, and 24.

Disease progress curves in unsprayed controls and those sprayed with mancozeb + sulfur at 14-day intervals were similar (Figures 22 and 24). The 7-day spray schedule of mancozeb produced progress curves with maximum disease severity below those of the other two spray schedules (Figure 23).

Vapam had no significant effect on nematode populations at preplant and harvest (Tables 36 and 37). There were virtually no nematodes detected at midseason.

Yield and gross dollar values per hectare are shown in Table 38. The highest yield and gross dollar values were obtained from fumigated plots with plants kept virtually disease free and not defoliated. The lowest yield was obtained from fumigated plots with plants which had 0.50 defoliation level and 0.71 disease severity (Table 38). Generally, yield responses were not proportional to defoliation levels and metam-sodium rates. Fumigation resulted in net loss of income regardless of defoliation level and disease severity (Table 39). No defoliation, 0.83 disease severity, and fumigation with vapam at 374 L/ha resulted in loss of income of $1191, $1320 and $1398 at the high, medium and low prices respectively whereas plots not fumigated, plants not defoliated, and having 0.87 disease severity had net gains in income of $300, $165 and $90 at the high, medium and low prices respectively (Table 39). Net gains in income varied in magnitude depending on disease severity and defoliation.

Discussion

The lack of consistent reduction in nematode populations on treatment with metam-sodium may be related to application technique.

There may have been less than adequate retention of the nematicide by the soil. This less adequate retention of the nematicide may have contributed to its apparent poor efficacy. Nematode populations were also generally low at all sampling times in the three tests.

In the study where metam-sodium alone was used, there was significant relationship between metam-sodium rates and yield. This significant difference may be due to metam-sodium controlling some soilborne plant diseases on beans since the nematicide had no significant effect on nematode populations and populations were very low.

Metam-sodium had no single or interactive effects on yield when it was used in combination with defoliation and/or bean rust in these tests. Defoliation was, however, the more important factor affecting yield when simultaneously used with metam-sodium. The non-significant interaction between defoliation and metam-sodium may be due to the generally low nematode populations on the site the study was conducted.

In the study where defoliation, metam-sodium, and bean rust were used simultaneously there were no significant interactions among the three factors. There were no significant differences in yield among defoliation levels, disease severity levels and metam-sodium rates. There were, however, significant differences in yield among fungicide sprays. Lack of interaction among the three factors may be due to low nematode populations and overriding effects of the disease. The disease was continuously associated with the crop from the time of inoculation to harvest. The overriding effects were shown by regression analysis which indicated that disease severity contributed most to the coefficient of determination. Thus, the continuous association of the disease with the crop may have influenced the physiology of the plants which

which was reflected in yield. The overriding effect of bean rust on yield was complicated by the fact that it was manipulated by fungicides which had micronutrients required for plant growth. Duncan's multiple range test showed that the micronutrient effect was not significant. To elucidate this micronutrient effect of fungicides on crop yield, further studies need to be conducted. Bitertanol may have controlled other pathogens not yet known, hence the higher yield.

Under the conditions these studies were conducted, the grower would have been better off not fumigating the soil due to the high cost of metam-sodium and low nematode populations. Fungicides, however, improved yield and hence gross income. The net return from fungicide sprays dpended on the frequency of spray. The optimum spray frequency was a 7-day schedule which indicated that spraying beans at intervals shorter than seven days was not beneficial. The loss a grower would incur depended on the price of snap beans and the cost of pesticides. Regression analysis appeared to be the best method of predicting yield, hence income, given various pest combinations. Regression analysis was able to show which factor contributed more to the coefficient of determination.

CHAPTER VII

THE EFFECT OF INOCULATION METHOD AND INITIAL POPULATION DENSITY OF MELOIDOGYNE INCOGNITA (KOIFOID AND WHITE) CHITWOOD ON SNAP BEANS (PHASEOLUS VULGARIS L.) 'SPRITE'

Introduction

The root-knot nematode Meloidogyne incognita (Kofoid and White) Chitwood poses a serious threat to bean (Phaseolus vulgaris L.) production in many bean growing areas of the world (Agudelo, 1980; Allen, 1983; Ngundo, 1977; Singh et al. 1981a; Sharma and Guazelli, 1982). Meloidogyne incognita infections have been reported to decrease the apparent photosynthetic rate of P. vulgaris 'Topnotch Golden Wax' as well as other physiological growth factors (Melakberhan et al., 1983). The limitation on bean production by root-knot nematodes may be due to root galling which interferes with nitrogen fixation by Rhizobium spp. and also interference with nutrient uptake. Yield losses of 50-90% have been reported from fields infested with root-knot nematodes (Agudelo, 1980; Freire and Ferraz, 1977; Ngundo, 1977; Varon and Galvez, 1974).

Damage functions ascribable to nematodes are influenced by many factors (McKenry, 1983). Nematode management decisions should, therefore, be based on environmental factors and the crops grown (Ferris, 1980). Environmental factors such as soil temperature, texture, and structure, and water infiltration rates influence moisture regimes of soil profiles which in turn affect nematode damage functions (McKenry, 1983). Noe and Barker (1983) related 24 edaphic variables to the field distribution of Meloidogyne spp. Work done on the influence of these

environmental factors on crop damage functions of root-knot nematodes has shown that these factors significantly affect the establishment of the nematodes and the growth of the crop and yield (Ferris, 1980; McKenry, 1983; Noe and Barker, 1983; Roberts, 1983).

The derivation of mathematical models relating nematode densities to crop damage has been discussed by Ferris (1980, 1984) and Seinhorst (1965, 1972). In these models, the relationship between the initial density of root-infesting nematodes and yield or other growth parameters of infected plants are expressed the assumptions that (i) up to a certain density the yield is not affected and (ii) a certain minimum yield remains unaffected by the nematodes even at the highest densities (Seinhorst, 1965).

The relationship between M. incognita and other Meloidogyne spp. initial population densities and plant growth and/or yield has been reported on tobacco (Barker et al., 1981, Ekanayake and Di Vito, 1984), beans (Melakberhan et al, 1983), tomato (Barker et al., 1976) and pepper (Di Vito et al., 1982). In these studies, inoculum consisted of eggs of M. incognita or other Meloidogyne spp. eggs extracted by the sodium hypochlorite method (Hussey and Barker, 1973) a factor which may be critical to the development of damage functions in these studies. Vrain (1977) evaluated the infectivity of three types of inocula which consisted of intact egg masses, eggs extracted with 0.53% NaOCl, and larvae hatched from NaOCl-treated eggs. The data obtained showed the limitation of egg masses, low infectivity from NaOCl-extracted eggs and sensitivity of larvae to relatively high temperatures.

Sodium hypochlorite dilutions have been used for the sterilization of the surface of nematodes and their eggs in laboratory studies

(Briggs, 1946; Feder and Feldmesser, 1955). Sodium hypochlorite has also been used for sterilizing processing substrates and equipment in diagnostic nematology laboratory work (Esser, 1972), so its adverse effects on nematodes at higher concentrations is well known.

The main objectives of this study were twofold (1) to determine the influence of the initial M. incognita population density on yield of beans and effects of inoculation method on the establishment of M. incognita on beans, and (2) to determine the effect of sodium hypochlorite (NaOCl) concentration on the number of M. incognita eggs and juveniles extracted from infected bean and tomato plants, and the influence of NaOCl concentration on egg hatch.

Materials and Methods

Meloidogyne incognita (Kofoid and White) Chitwood, obtained from Hausa potato (Coleus parviflorous Benth.), was maintained on greenhouse-grown tomato (Lycopersicon esculentum Mill 'Floradade'). Infected bean roots were obtained from an earlier experiment conducted in a greenhouse at the Tropical Research and Education Center in Homestead, Dade County, Florida. Sodium hypochlorite (NaOCl) solutions (0.13, 0.26, 0.525, 1.3 and 2.6%) were made from Thrift King(R) commercial bleach (5.25% NaOCl) diluted serially with cold tap water (25°C). Roots were cleaned of soil, cut into 2-3 cm pieces, mixed, and 120 g of the mixture was used for egg and juvenile extraction by the sodium hypochlorite method (Hussey and Barker, 1973), except that a 230-mesh sieve was used instead of the 200-mesh. Comparisons of egg and juvenile numbers extracted by the various concentrations of NaOCl were made.

For the egg-hatch test, 1 ml of M. incognita eggs and juveniles suspended in tap water was put in a watch glass and 2 ml of tap water was added to the suspension. The initial number of eggs and juveniles was determined by counting them under a dissecting microscope. The watch glasses were kept at room temperature (24-30°C). Subsequent egg hatch in each treatment were assessed every 2 days until hatching levelled off (Vrain (1977).

The inoculation method studies were established on 31 August 1984, in 1-quart side-drain black plastic pots filled with 1 L of soil (1 part sand to 3 parts Palmetto Rich Earth(R). The soil was inoculated with eggs and juveniles extracted with the 0.525% NaOCl solution. One series of pots was inoculated by thoroughly mixing the inoculum with the soil. The other series was inoculated by drenching the seeds with the inoculum. A third series was inoculated by placing the appropriate number of galls in the pot. Each gall contained an average of 246 eggs and juveniles.

The egg and juvenile populations investigated were 0, 10, 100, 1,000, 10,000, and 100,000 per pot and the numbers of galls were 0, 1, 10, 100, and 500 galls per pot. Since each gall contained an average of 246 eggs and juveniles the gall inoculum was, therefore, equivalent to 0, 246, 2,460, 24,600 and 123,000 eggs and juveniles per pot. Three seeds were planted in each pot and plants were thinned to one plant/pot after germination. Treatments were replicated four times in a randomized complete block. Pots were placed on corrugated benches 0.91 m high in an open greenhouse and watered twice daily using an automatic time-controlled water mist-forming system. Beans were harvested on 25

October 1984 and root gall indices determined following the method outlined by Taylor and Sasser (1978).

Yield data were subjected to regression analysis using the general linear models of SAS. Seinhorst model curve fitting was also attempted.

Results

The relationship between NaOCl concentration and $\log_{10}$ (number of eggs and juveniles + 1) extracted is shown in figure 29. Data fit the equations $Y = a + b\ LnX$ or $Y = a\ X^b$ where Y = log (number of eggs and juveniles + 1), X = concentration (%) of NaOCl, and a and b are constants. The coefficient of determination (R^2) values were 0.69* to 0.75* for the bean and tomato curves, respectively. Figure 29 shows that the number of eggs and juveniles extracted increased more rapidly at low NaOCl concentrations (0 to 0.26%) and levels off at high concentrations (0.525-2.6%).

Table 40 shows the number of Meloidogyne incognita eggs and juveniles extracted form 120 g of infected plant roots at various NaOCl concentrations. These eggs and juveniles were extracted from bean and tomato roots 63 and 50 days after inoculation respectively.

Figures 30 and 31 show the total number of juveniles which emerged from eggs extracted at various NaOCl concentrations. On the day of extraction, sodium hypochlorite-extracted eggs hatched more than the water-extracted eggs. The highest number of juveniles had emerged from eggs extracted with the 0.525% NaOCl solution from bean roots over the next 6 days after extraction (Figure 30). Eggs extracted from tomato roots had a different hatch trend from that of beans (Figure 31). The

0.525% solution gave the best overall hatch of nematode eggs extracted from both bean and tomato roots (Figure 30 and 31).

The percentage hatch of M. incognita eggs is shown in figure 32. The lowest percentage hatch was obtained from the 2.6% NaOCl-extracted eggs. Water-extracted eggs had a percentage hatch comparable to that of the 0.13, 0.26 and 0.525% NaOCl-extracted eggs. The 0.525% NaOCl-extracted eggs had the highest proportion of eggs hatched among all treatments by day 8, leading to the choice of the 0.525% NaOCl-extracted eggs in the inoculation of bean plants and maximum yield of eggs over time as above.

The relationship between initial M. incognita densities and snap bean yield is shown in figure 33. There was a significant ($P = 0.01$) negative correlation between nematode densities and yield for all three inoculation systems (correlation coefficients were -0.96, -0.78 and -0.96 for seed drench, soil mix and gall inoculation). As the nematode densities increased, the snap bean yield decreased. The lowest yield was obtained from plants inoculated with 500 galls/pot (= 123,000 eggs/pot). Negative impacts on yield were greater in seed-drench inoculated pots than soil-mix inoculated ones (Figure 33). There were significant differences in yield among nematode densities in the seed-drench and gall inoculation methods with F values of 14.34** and 26.18**, respectively. There were, however, no significant differences in yield among nematode densities in the soil-mix inoculation method with an F value of 3.09.

Gall indices were comparable in all three inoculation methods as shown in Table 41. Among inoculated plants, the lowest gall index was observed on plants inoculated with 10 eggs and juveniles per pot.

TABLE 40. Total number of eggs and juveniles extracted from 120 g of bean and tomato roots. Data are means of 3 replicates.

NaOCl Concentration	Inoculum Source	
(%)	Bean	Tomato
0	135,000	320,000
0.13	290,000	704,000
0.26	517,000	1,566,000
0.525	642,000	2,120,000
1.3	864,000	2,886,000
1.6	1,062,000	3,228,000

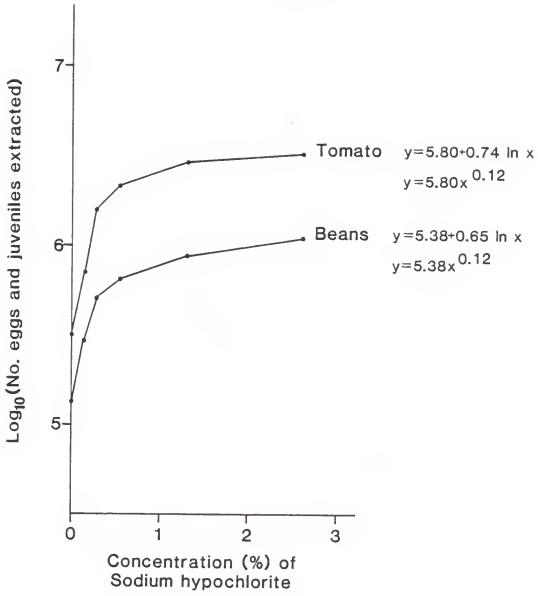


Figure 29. Log_{10}(No. eggs and juveniles extracted) at various NaOCl concentrations.

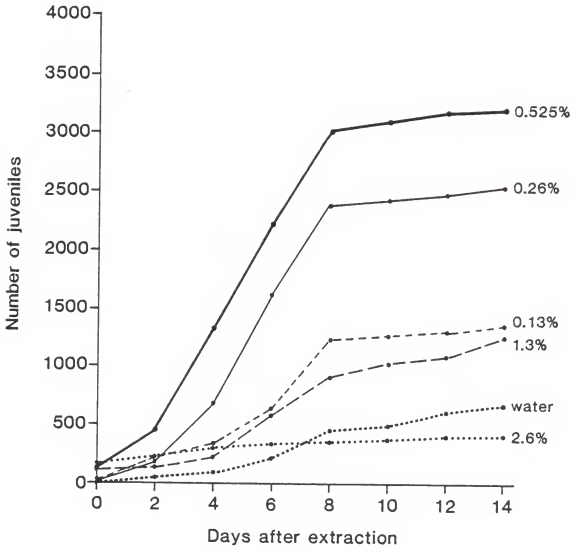


Figure 30. No. juveniles hatched from eggs obtained from bean plants.

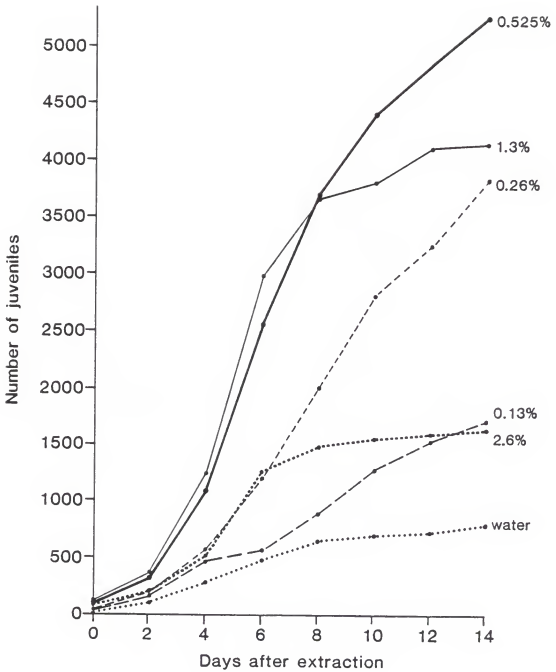


Figure 31. No. juveniles hatched from eggs obtained from tomato plants.

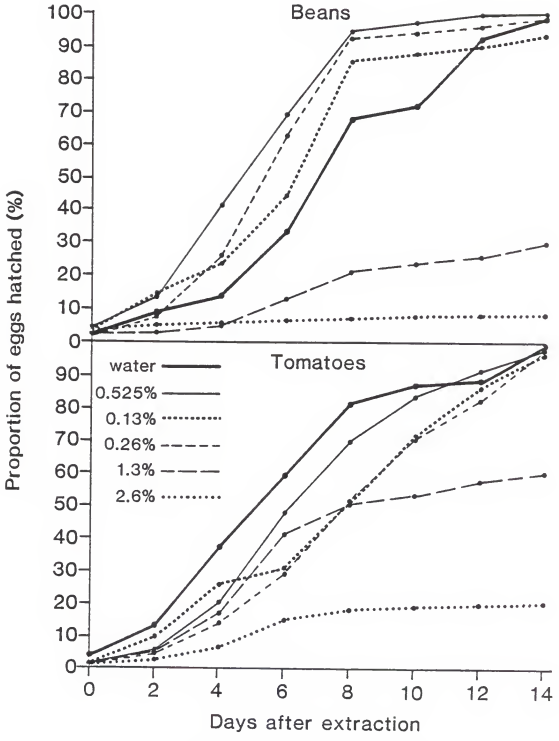


Figure 32. Egg hatch (%) at various NaOCl concentrations.

TABLE 41. Yield and root gall indices of bean plants treated with low to high initial densities of Meloidogyne incognita eggs and juveniles. Data are means of 4 replicates.

Mean initial number of eggs and juveniles/1L soil		Yield (g/plot)	Gall indices[a] (means)
Untreated check		19	0
10	(Seed drench)	18	2
100	" "	14	4
1,000	" "	13	5
10,000	" "	11	5
100,000	" "	4	5
10	(Soil mix)	12	3
100	" "	15	4
1,000	" "	15	4
10,000	" "	8	5
100,000	" "	9	5
246	(1 gall)	13	5
2,460	(10 galls)	4	5
24,600	(100 galls)	1	5
123,000	(500 galls)	1	5

[a] Gall index data based on the scale of Taylor and Sasser (1978) as follows: no galls or egg masses = 0; 1-2 galls or egg masses = 1; 3-10 = 2; 11-30 = 3; 31-100 = 4; more than 100 = 5.

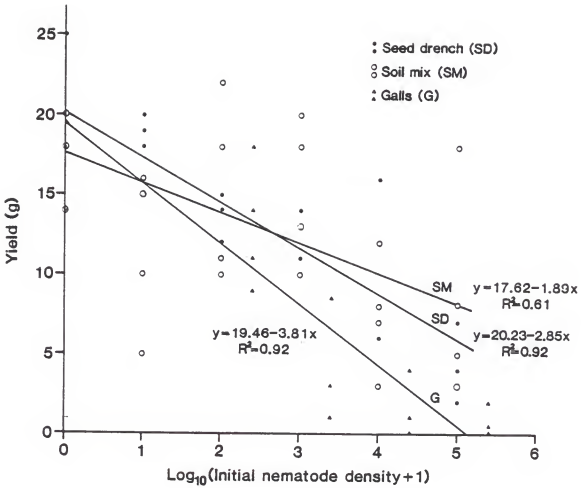


Figure 33. Relation between $\log_{10}$ (initial nematode density +1) and yield.

Plants inoculated with one gall/pot had a similar gall index to that of the plants inoculated with 100 or 1,000 eggs and juveniles/pot as anticipated. Ten galls per pot gave gall indices similar to these of 10,000 or 100,000 eggs and juveniles/pot. Plants inoculated with more than 10 galls gave the maximum gall index. The controls had no root galls.

Data obtained in this study did not fit the Seinhorst model (Seinhorst, 1965, 1972) which is of the form $Y = M + (1-M)Z^{[P-T]}$ where Y = ratio between the yield at nematode population P and at $P < T$, M = relative minimum yield, P = initial nematode population density, T = tolerance limit for the nematode density, and Z = a constant. Coefficients of determination were 0.031, 0.003, and 0.28 for the soil mix, seed drench, and gall inoculation systems. Linear regression analysis of yield data is shown in Figure 33. The linear model produced significant coefficients of determination at the 0.01 probability level.

Discussion

The largest number of M. incognita eggs and juveniles was extracted with the 2.6% NaOCl solution, due to the better dissolution of the gelatinous matrix enclosing the nematode eggs at the highest NaOCl concentration tested. The rate of increase of eggs and juveniles extracted, however, decreased dramatically above the 0.525% concentration probably because the number of egg masses or galls per sample was the same. Further the increase in NaOCl did not improve egg and juvenile extraction that much.

There were more eggs and juveniles extracted from tomato than from bean roots (Table 40). This may be an indication that tomato is a more comparitible root-knot nematode host than beans. Consequently the nematodes may have reproduced at a faster rate on tomato than on beans. It is possible that this difference in extracted eggs and juveniles may be a reflection of age differences. Eggs and juveniles were extracted 63 days after inoculation from bean roots whereas eggs and juveniles were extracted from tomato plants 50 days after inoculation. Thus, there might have been more juveniles which had already emerged from bean root galls than on tomato root galls. Consequently, more juveniles may have been washed off along with soil from bean roots which may have lead to the low numbers of eggs and juveniles extracted. Moreover, weighing did not imply the same number of galls or eggs on the root material used. Weighing may thus have biased the number of egg masses used towards the tomato source. There is little information in the literature pertaining to variation in the number of eggs per gall in various hosts. This suggests that variation in egg numbers per gall on various hosts be investigated.

More juveniles of M. incognita may have emerged on the day of extraction from NaOCl-treated eggs than from water-treated eggs, probably because the NaOCl reacted with the egg shell, thereby inducing early hatch in eggs with fully developed juveniles. This effect of NaOCl on egg hatch had, however, some negative effects when the concentration was above 0.525%. The negative effect of NaOCl on eggs was particularly evident with the 2.6% NaOCl treated eggs. This high concentration of NaOCl gave rise to numbers of juveniles comparable to those obtained from water-treated eggs, despite the 2.6% solution having extracted the

highest number of eggs from either crop. The 0.525% solution had the highest number of juveniles that emerged from eggs obtained from either beans or tomato. Thus, a 0.525% NaOCl solution may be the optimum concentration for M. incognita egg extraction for inoculation studies. The low emergence of juveniles from water-treated eggs may imply that under natural conditions root-knot nematode eggs hatch over a long period. This may be a survival mechanism for this nematode.

There were no significant differences in the proportion of eggs that had hatched between water and NaOCl solutions of 0.13, 0.26 and 0.525%. This was because there were far fewer eggs extracted with water than with the NaOCl treatments. The lowest percentage hatch was obtained from eggs extracted with 2.6% NaOCl. This low hatch was probably due to the adverse effects of high NaOCl concentration on eggs which may have arrested the development of embryos of juveniles.

Gall inoculations had a more marked effect on yield and gall indices (as evidenced by greater slope) than extracted eggs probably because eggs in intact egg masses hatched in their natural environment and there was no loss from mortality due to NaOCl. The effect of egg extraction may have been exacerbated by the inoculation of soil before seeds germinated. Thus, juveniles may have been rendered less infective before seeds germinated. Root-knot nematodes are more infective at the second juvenile than any other stage (Dropkin, 1980). Seeds germinated 5 days after planting and nematode inoculation. A good proportion of eggs may have hatched well before the plants produced roots. Nematode juveniles, especially root-knot nematodes, are sensitive to relatively high edaphic temperatures (McKenry, 1983). Root gall indices were

comparable in all three inoculation methods. The extent of galling was, however, not reflected in yield in the soil-mix inoculated plants.

There were significant differences in snap bean yield among nematode egg and juvenile densities in the seed-drench method ($F = 14.34$, $P \leq 0.01$). This may be due to some juveniles ecloding from the eggs in the vicinity of the germinating seeds since the nematode eggs hatched over a period of up to 14 days. Hence many of them may have been able to penetrate bean radicles soon after hatching. There were no significant differences in snap bean yield ($F = 3.09$, $P \leq 0.1$) among nematode egg and juvenile levels in the soil-mix inoculation method probably because some of the eggs and juveniles leached out with irrigation water. Moreover, juveniles may not have been able to swim up to the vicinity of germinating seeds against the downward flow of water. The seed drench method apparently concentrated nematodes, more, within the vicinity of the plant root system so the local population density was higher than if much of the inoculum was scattered through the entire volume of soil. Gall-inoculated plants produced the lowest yield and differences in yield among gall treatments were significant at $P = 0.01$ ($F = 26.18$). This may be due to the natural environment in the galls within which juveniles ecloded over a long period.

There was lack of association between yields obtained from seed-drench and soil-mix inoculated plants ($\chi^2 = 7.74$, $0.10 < P < 0.20$). This lack of association does not necessarily prove that the yields were different from each other but there was a chance that they were different.

The data did not fit the Seinhorst model probably because the nematode population levels were in a logarithmic progression. In most

of the work in which this model was able to explain the relationship between yield and nematode population densities, the intervals between population levels were smaller particularly at low densities. Thus, where nematode population levels are far apart, the Seinhorst model may not be the best for explaining the relationship between plant growth parameters and nematode population levels.

Results presented here indicate that M. incognita may cause yield loss in snap beans regardless of the method of inoculation. If the soil is mixed with 0.525 NaOCl-extracted eggs the influence on yield may be less profound than if the plants were inoculated with galls or via seed drench. It has, also, been shown that NaOCl concentrations have a direct positive relationship to the numbers of nematode eggs and juveniles extracted. Indicated in the results is the fact the NaOCl has an egg-hatch inducing effect, compared to eggs allowed to stand in tap water at room temperature, which is less pronounced as the concentration increases above 0.525%.

CHAPTER VIII
SUMMARY AND CONCLUSIONS

Pest damage and disease infection on beans rarely result in total leaf abscision under field conditions. In the experiments conducted in this study, various defoliation levels up to total defoliation were included. Bean rust was manipulated by fungicide sprays at various frequencies and nematode infestation was studied under field as well as greenhouse conditions. These factors were studied individually and in combination.

Bean plants were most sensitive to defoliation at full-bloom and pod-set growth stages. Twenty five percent leaf removal at the primary leaf and first trifoliate leaf stages resulted in yield losses of up to 36% and 29% in the greenhouse and field respectively. In the greenhouse 50%, 75%, and 100% defoliation resulted in up to 34%, 39%, and 74% yield loss, respectively. In the field 50%, 75%, and 100% defoliation caused yield losses of up to 68%, 70%, and 95% respectively depending on the plant growth stage. Yield loss fluctuated in such a way that in some cases lower defoliation levels resulted in higher yield losses than higher defoliation levels. This fluctuation may have been due to various factors, one of which could be better exposure of photosynthetically active foliage to light.

Root-knot nematodes, Meloidogyne incognita, caused yield loss in snap beans when plants were inoculated with eggs and juveniles. Ten eggs and juveniles per pot resulted in 19% yield loss and 100, 1,000, 10,000, and 100,000 eggs and juveniles resulted in 38%, 45%, 47%, and

59% yield loss. Thus, the threshold level of M. incognita was between 0 and 10 eggs and juveniles per pot. Combining defoliation and root-knot nematodes showed that there was no significant interaction between them. When defoliation was held constant, yield reduction rate was greater than when root-knot nematode populations were held constant. Equations were devised to express this relationship between the two factors.

Bean rust caused yield loss in snap beans, which was substantial at the highest disease severity (maximum proportion of foliage infected). In the first trial, disease severity levels of 0.098, 0.46, 0.65, and 0.76 caused yield losses of 21%, 17%, 35%, and 60% respectively. Disease severity levels of 0.4, 0.47, 0.71, and 0.86 resulted in yield losses of 55%, 56%, 80%, and 91% in trial 2. Thus, the disease severity level which could be tolerated was below 0.1. When bean rust was used simultaneously with defoliation and nematodes, it was demonstrated by regression analysis of the data that the disease had an overriding effect on yield loss. The disease may have had this overriding effect due to its continual adverse effects on the plants from its onset to bean harvest. In these experiments, nematodes and defoliation had little effect on yield. There was no significant interaction among nematodes, defoliation and bean rust. As expected the best yield was obtained from plants free of all stress.

From these results, several general conclusions can be drawn. Some defoliation may result in exposure of photosynthetically active foliage to light and also may stimulate rapid growth of new leaves which are highly photosynthetically active. Yield reduction due to defoliation depends on the growth stage at which leaf removal occurs. This may be due to the differences in partitioning of photosynthates at these various growth stages.

The nematode, M. incognita, caused substantial yield loss when plants were inoculated in pots, but hardly had any effect on yield under field conditions. The lack of apparent effect on yield by nematodes may be due to interaction of several biotic as well as abiotic factors in field soils. Moreover, the nematode populations in the field were very low, likely were below threshold levels. In the greenhouse, plants were grown in pots in which nematodes were probably concentrated on the upper soil layer thus making the nematode density higher. This discrepancy between nematode effects on yield in the greenhouse and field may be an indication that threshold levels determined in the greenhouse may not necessarily apply under field conditions.

Bean rust caused apparent yield loss even at the lowest maximum severity level measured. This may be due to the continual adverse effects of the disease on the physiology and other bean plant growth factors. A low disease severity, however, required the use of mancozeb + sulfur more often hence increasing production costs. Increasing production costs in turn reduced net returns from the crop. The improvement in yield by mancozeb + sulfur may also have been due to increased supply of micronutrients, such as zinc, which stimulated plant growth. Thus, yield increases in beans may not necessarily be due to bean rust control/prevention only.

There may not necessarily be a close relationship between experimental results obtained in the greenhouse and the field. Thus, pest control recommendations should not be based only on greenhouse/laboratory experimental results. Recommendations for control of a pest complex should be based on the population dynamics of the key pests supplemented by the presence of other factors. Pest population levels can be obtained by close and routine population monitoring throughout the growing season.

Yield was generally low in these experiments, probably because of insect pest pressure, especially cowpea curculio and other leaf-feeding insects. In the defoliation, metam-sodium, and bean rust disease experiment the low yields may have been due to the late planting thus going into a season where rainfall was erratic and insect pests were more abundant. Generally, the weather conditions were not optimum for snap bean production during the period some of the field experiments were conducted.

A multipest assessment on beans in the field was not easy because it was time consuming and tedious. Thus, a grower/extension worker would have to assess pests separately and act on them accordingly. This has been shown to be a valid approach in these experiments by the general lack of interaction among defoliation, nematodes or nematicide, and bean rust disease.

APPENDIX
EFFECTS OF DEFOLIATION AND FUNGICIDES ON SNAP BEANS

Introduction

Bean rust, caused by Uromyces phaseoli (Pers.) Wint., is one of the most prevalent diseases on beans (Phaseolus vulgaris L.) grown in Florida and other bean producing areas of the world (Agudelo, 1980; Allen, 1983; Cook, 1978; Kidney, 1980). Bean rust is common in warm, (15-27°C) moist weather (Augustin et al., 1972; Gonzalez, 1976; Shurtleff, 1966). Uromyces phaseoli is an autoecious/monoecious fungus which infects mainly plants in the legume family (Cook, 1978; Shurtleff, 1966). Symptoms of the disease are confined largely to leaves, but young stems and branches are occasionally infected. On severe infection symptoms or signs may appear on pods (Cook, 1978; Vargas, 1980). Lesions first appear as small slightly raised spots that are light in color and more often on the lower leaf surfaces. These pustules gradually enlarge to 1-2 mm in size and may rupture the leaf epidermis 8 days after infection (Allen, 1983). A ring of secondary sori usually develops around the original infection locus on susceptible varieties.

Bean rust is commonly controlled by weekly sprays of mancozeb tank-mixed with sulfur or other of the ethylene bis-dithiocarbamate fungicides (Pohronezny et al., 1984). A few experimental fungicides are available for bean rust control. One of these more potent fungicides for bean rust control is bitertanol. Thus, this study was conducted to

investigate the effect of manual defoliation and bitertanol and mancozeb sprays at various frequencies on bean rust and snap bean yield, the original objective of which had been to determine the effects of bean rust disease and defoliation on yield. However, due to lack of disease development, the test was limited to evaluation of the direct effects of the fungicides and defoliation on snap bean yield.

Materials and Methods

Soil was fumigated with Dowfume MC2(R) at a rate of 314 kg/ha. Two-gallon side drain plastic pots were filled with 6.4 L of soil and placed in the field. A plot consisted of 2 pots each with 3 plants. Plots were 1.5 m apart. Treatments were replicated 4 times in a randomized complete block. Treatments were arranged in a 5x5x2 factorial design. Fertilizer (8:16:16) was applied at 448 kg/ha (to pots and incorporated) and plants were top dressed at 224 kg/ha just before flowering. Trigard(R) (150g/ha) was applied for leafminer control. Mesurol was applied for slug and snail control as needed. Other leaf-eating insects were controlled with sprays of permethrin or endosulfan.

Beans (Phaseolus vulgaris L., 'Sprite') were planted on 25 January 1985 and harvested on 25 April 1985. Irrigation was provided by an overhead sprinkler system twice a week. Plants were defoliated at two growth stages, the primary leaf stage and flower bud formation. Defoliation levels investigated were 25%, 50%, 75%, and 100% plus an undefoliated control. The foliage was removed from the distal end of the petiole using a pair of scissors.

Two fungicides were tested for the manipulation of bean rust. These were; bitertanol (57 g ai/ha) applied at 7-day intervals, and mancozeb (1.7 kg/ha) tank-mixed with sulfur (4.2 kg ai/ha) applied at 4-5, 7, and 14-day intervals. An unsprayed control was also included.

Inoculum exposure was done at the primary leaf stage by clipping infected pole bean leaves on a string tied across experimental plots 1.5 m above the ground. Fungicide sprays were initiated soon after inoculation. Disease progress was assessed by the Horsfall-Barratt rating system.

Data were subject to analysis of variance.

Results

Analysis of variance on the effect of fungicides and defoliation on snap bean yield showed that there were significant differences among fungicides and manual defoliation levels ($F = 18.32^{**}$ and 29.34^{**} ($P \leq 0.01$) respectively) (Table 42-1). There were, however, no significant interactions between fungicides and manual defoliation levels (Table 42-1). There were no significant differences between defoliation times ($F = 1.39$ N.S.) and interaction among manual defoliation levels and time ($F = 1.09$ N.S.). Duncan's multiple range test showed that there were no significant differences in yield between the unsprayed plants and those sprayed with mancozeb and sulfur at 7 and 14-day intervals (Table 42-2). There were, however, significant differences in yield between mancozeb at all schedules and bitertanol (Table 42-2).

The highest snap bean yield was obtained from plots sprayed weekly with bitertanol at all defoliation levels and both times of defoliation

TABLE 42-1. F-values and probabilities from analysis of variance on the effects of defoliation, fungicides and their interaction on snap bean yield.

Source	F	Probability F
Defoliation	29.34	0.0001
Time	1.39	0.24
Defoliation x Time	1.09	0.36
Fungicide	18.32	0.0001
Defoliation x Fungicide	0.74	0.75

TABLE 42-2. Mean snap yield per hectare sprayed with various fungicides.

Fungicide	Spray frequency (days)	Mean yield (g/plot)
No fungicide	0	114 cd
Mancozeb + sulfur	14	98 d
Mancozeb + sulfur	7	119 c
Mancozeb + sulfur	4-5	137 b
Bitertanol	7	166 a

Means with the same letter are not significantly different at $P \leq 0.05$ (Duncan's multiple range test).

TABLE 43. Mean snap bean yield from plants defoliated at various levels and various fungicide spray frequencies.

Defoliation levels (%)	Fungicide	Plant Growth Stage	
		Primary leaf	Flower bud formation
0	No fungicide	143	152
0	Bitertanol (7-day)[a]	212	232
0	Mancozeb (4-5-day)	185	157
0	Mancozeb (7-day)	144	155
0	Mancozeb (14-day)	126	131
25	No fungicide	112	126
25	Bitertanol (7-day)	181	172
25	Mancozeb (4-5-day)	162	152
25	Mancozeb (7-day)	147	149
25	Mancozeb (14-day)	116	73
50	No fungicide	99	95
50	Bitertanol (7-day)	180	168
50	Mancozeb (4-5-day)	154	153
50	Mancozeb (14-day)	106	128
75	No fungicide	104	98
75	Bitertanol (7-day)	135	134
75	Mancozeb (4-5-day)	121	100
75	Mancozeb (7-day)	108	106
75	Mancozeb (14-day)	109	59
100	No fungicide	100	74
100	Bitertanol (7-day)	93	106
100	Mancozeb (4-5-day)	81	113
100	Mancozeb (7-day)	60	77
100	Mancozeb (14-day)	76	56

[a] Figures in parentheses are spray intervals.

TABLE 44. Effect of fungicides and defoliation on gross dollar values per hectare of snap beans.

Defoliation level (%)	Fungicide	Price range	Plant Growth Stage of defoliation: Primary leaf	Plant Growth Stage of defoliation: Flower bud formation
0	No fungicide	low	1122	1193
		medium	2054	2186
		high	3740	3975
0	Bitertanol (7-day)	low	1663	1820
		medium	3049	3337
		high	5544	6067
0	Mancozeb (4-5-day)	low	451	1232
		medium	2661	2258
		high	4838	4106
0	Mancozeb (7-day)	low	1130	1216
		medium	2071	2230
		high	3766	4054
0	Mancozeb (14 days)	low	989	1028
		medium	1812	1884
		high	3295	3426
25	No fungicide	low	879	973
		medium	1611	1784
		high	2929	3243
25	Bitertanol (7-day)	low	1420	1349
		medium	2604	2474
		high	4734	4498
25	Mancozeb (4-5-day)	low	1271	1193
		medium	2330	2186
		high	4237	3975
25	Mancozeb (7-day)	low	1153	1169
		medium	2114	2143
		high	3844	3897
25	Mancozeb	low	1028	973
		medium	1884	1784
		high	3426	3243
50	No fungicide	low	777	745
		medium	1424	1366
		high	2589	2485

TABLE 44. Continued

Defoliation level (%)	Fungicide	Price range	Plant Growth Stage of defoliation: Primary leaf	Plant Growth Stage of defoliation: Flower bud formation
		low	1412	1318
50	Bitertanol (7-day)	medium	2589	2417
		high	4707	4393
		low	1208	1200
50	Mancozeb (4-5-day)	medium	2215	2201
		high	4028	4001
		low	842	1004
50	Mancozeb (7-day)	medium	1544	1841
		high	2807	3348
		low	910	573
50	Mancozeb (14-day)	medium	1669	1050
		high	3034	1909
		low	816	769
75	No Fungicide	medium	1496	1410
		high	2720	2563
		low	1059	1051
75	Bitertanol (7-day)	medium	1942	1927
		high	3531	3504
		low	949	785
75	Mancozeb (4-5-day)	medium	1740	1438
		high	3164	2615
		low	847	832
75	Mancozeb (7-day)	medium	1568	849
		high	2824	2772
		low	855	463
75	Mancozeb (14-day)	medium	1568	849
		high	2851	1543
		low	785	581
100	No Fungicide	medium	1438	1064
		high	2615	1935
		low	730	832
100	Bitertanol (7-day)	medium	1338	1525
		high	2432	2772

TABLE 44. Continued

Defoliation level (%)	Fungicide	Price range	Plant Growth Stage of defoliation: Primary leaf	Plant Growth Stage of defoliation: Flower bud formation
		low	636	887
100	Mancozeb (4-5-day)	medium	1165	1625
		high	2118	2955
		low	471	604
100	Mancozeb (7-day)	medium	863	1108
		high	1569	2014
		low	596	439
100	Mancozeb (14-day)	medium	1093	806
		high	1988	1465

TABLE 45. Net income (dollars) per hectare of snap beans defoliated at the primary leaf and flower bud formation stages and sprayed with various fungicides

Defoliation level(%)	Fungicide	Plant growth stage: Primary leaf	Plants were defoliated: Flower bud formation
0	No fungicide	0	0
0	Bitertanol (7-day)	1805	2092
0	Mancozeb (4-5-day)	929	- 39
0	Mancozeb (7-day)	- 87	- 34
0	Mancozeb (14-day)	- 501	- 662
25	No fungicide	- 811	- 732
25	Bitertanol (7-day)	694	523
25	Mancozeb (4-5-day)	328	- 169
25	Mancozeb (7-day)	- 8	191
25	Mancozeb (14-day)	- 370	- 789
50	No fungicide	-1151	-1491
50	Bitertanol (7-day)	968	418
50	Mancozeb (4-5-day)	118	- 143
50	Mancozeb (7-day)	-1046	- 741
50	Mancozeb (14-day)	- 763	-2123
75	No fungicide	-1020	-1412
75	Bitertanol (7-day)	- 209	- 471
75	Mancozeb (4-5-day)	- 745	-1529
75	Mancozeb (7-day)	-1028	-1316
75	Mancozeb (14-day)	- 946	-2489
100	No fungicide	-1125	-2040
100	Bitertanol (7-day)	-1308	-1203
100	Mancozeb (4-5-day)	-1791	-1189
100	Mancozeb (7-day)	-2284	-2074
100	Mancozeb (14-day)	-1809	-2567

except for total defoliation (Table 43). The unsprayed plots gave yields comparable to those obtained from fortnightly mancozeb-sprayed plants. There was no apparent difference between the 4-5-day and 7-day mancozeb spray schedules (Table 43).

Gross dollar values per hectare of snap beans are shown in Table 44. Bitertanol-sprayed plots consistently gave the highest dollar values at all defoliation levels and both growth stages. The gross dollar values were computed from the following price range, $6.00 (low), $11.00 (medium), and $20.00 (high) per bushel (13.62 kg/) of snap beans multiplied by the yield per hectare. Net income (Table 45) was derived from the no spray value as the base line. This was deducted from the values of the other treatments and the cost of fungicide sprays deducted from this difference. Bitertanol is an experimental fungicide which has not been registered for use on beans, hence the price is not known. Thus, the net value of beans for this fungicide excludes its cost. Consequently, the net values for bitertanol may not be a true reflection. Under the conditions this study was conducted, the grower would have made a profit if he sprayed his crop with mancozeb at 4-5 day intervals even if 35% or 50% of the leaf area were removed at the primary leaf stage (Table 45). There was no positive effect on net income from fungicide sprays if plants were defoliated at the flower bud formation stage.

Discussion

There were significant differences among fungicide spray schedules based on yield. Bitertanol gave the highest yield and by derivation the

largest dollar values. Four to five-day sprays of mancozeb were generally better than 7-day spray schedules on yield. Bitertanol gave the highest yield and highest dollar values. Four to five-day sprays of mancozeb and sulfur were generally better than 7-day sprays. This may have been due to the presence of zinc, an element required for plant growth. May be the more frequent sprays provided more of this element than the 7-day spray schedule of mancozeb and sulfur rather than the efficacy of the fungicide since there was virtually no apparent disease build up on the plants. It is possible that the more frequent sprays controlled other diseases resulting in higher yield. This same fact may be true for the fortnightly spray schedule which generally gave the lowest yield among the fungicide sprays.

The net income value picture is not clear with regard to bitertanol since its market price was not known. Moreover, the net income depends on snap bean prices which fluctuate widely. Furthermore, these values do not include other production costs such as farm machinery, labor and interest on loans. There was no apparent benefit from the shorter (4-5 day) spray schedule. The extra sprays did not produce enough increase in yield to justify added cost. From these results the ideal mancozeb spray schedule was the 7-day interval. Biteranol increased snap bean yield substantially probably because this chemical controlled some other subtle disease and it may have plant growth regulating properties. This has been suggested by teh non-establishment of bean rust disease in this study. Bitertanol has no known micronutrients which ruled out the micronutrient effect.

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BIOGRAPHICAL SCKETCH

Afete Divelias Gadabu was born on 19 March 1949, in Lilongwe, Malawi. He attended primary school at Kamzimbi Primary School and went to Bwaila Secondary School where he got his Junior Certificate of education in 1968. He went to Blantyre Secondary School in 1969 and obtained a Cambridge School Certificate in 1970. In 1971, he received a scholarship to the University of Malawi from which he graduated with a Bachelor of Science in biology and chemistry in 1975.

In the same year he joined the Civil Service as a research officer in the Department of Agricultural Research of the Ministry of Agriculture. In 1976 he received a scholarship to the University of Newcastle upon Tyne, U.K. He graduated with a Master of Science in applied entomology in 1978 and returned to Malawi. He worked for the Department of Agricultural Research as a research entomologist. In 1982, he received a scholarship to the University of Florida where he has been a graduate student in the Department of Entomology and Nematology from 1983 to 1986. Upon fulfilling the requirements for the degree of Doctor of Philosophy, he is returning to Malawi to continue working as a research entomologist.

I certify that I have read this study and that in my opinion it conforms to acceptable standards of scholarly presentation and is fully adequate, in scope and quality, as a dissertation for the degree of Doctor of Philosophy.

V. H. Waddill, Chairman
Professor of Entomology
and Nematology

I certify that I have read this study and that in my opinion it conforms to acceptable standards of scholarly presentation and is fully adequate, in scope and quality, as a dissertation for the degree of Doctor of Philosophy.

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May 1986

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